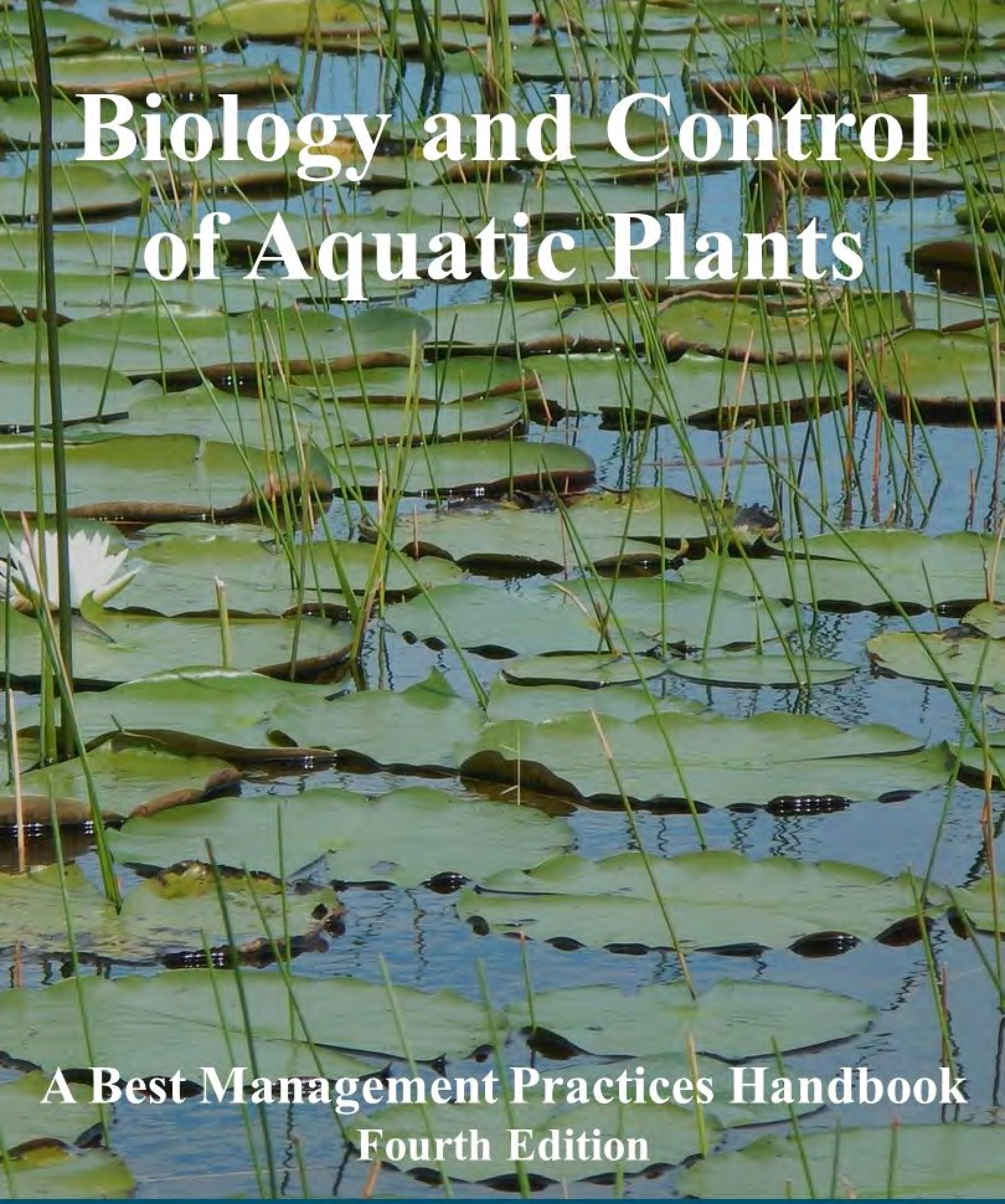


Biology and Control of Aquatic Plants

A Best Management Practices Handbook

Fourth Edition

Lyn A. Gettys, William T. Haller and David G. Petty, editors

July 2020

Dear Reader:

Thank you for your interest in aquatic plant management. The Aquatic Ecosystem Restoration Foundation (AERF) is pleased to bring you the fourth edition of *Biology and Control of Aquatic Plants: A Best Management Practices Handbook.*

The mission of the AERF, a not for profit foundation, is to support research and development which provides strategies and techniques for the environmentally and scientifically sound management, conservation and restoration of aquatic ecosystems. One of the ways the Foundation accomplishes the mission is by providing information to the public on the benefits of conserving aquatic ecosystems. The handbook has been one of the most successful ways of distributing information to the public regarding aquatic plant management. The first, second and third editions of this handbook became some of the most widely read and used references in the aquatic plant management community. This fourth edition has been specifically designed with water resource managers, water management associations, homeowners and customers and operators of aquatic plant management companies and districts in mind. It is not intended to provide the answers to every question, but it should provide basic scientifically sound information to assist decision-makers.

The authors, editors and contributors reflect the best the aquatic plant management industry has to offer. They gave generously of their time and talent in the production of this handbook and they deserve all the praise and thanks that can be garnered. Not only have they prepared the sections, they are available to all interested parties to provide clarification and additional information as warranted. These scientists, professors, aquatic plant managers and government officials have created a document that surely will be the most widely read and circulated handbook produced to date. Thank you all.

The production of this document has been made possible through the generosity of sponsors of the Foundation. I offer my thanks and appreciation to these faithful supporters who continue to underwrite what has been an effort to provide the very best handbook possible.

I hope you find this handbook to be helpful and informative. A downloadable version is on the AERF website at **www.aquatics.org** along with other useful information and links. Consider becoming a sponsor of the Foundation and supporting educational projects and other ecosystem restoration efforts across the country.

Carlton R. Layne
Executive Director

Restoration Regulatory Research

Contributors

Marc Bellaud (mbellaud@solitudelake.com)
SOLitude Lake Management
590 Lake Street, Shrewsbury MA 01545

Brett Bultemeier (bwbult@ufl.edu)
University of Florida IFAS Pesticide Information Office
7922 NW 71st Street, Gainesville FL 32653

Gregory M. Chorak (gregory.chorak@student.montana.edu)
Montana State University Plant Sciences and Plant Pathology Department
119 Plant Biosciences Building, Bozeman MT 59717

James Cuda (jcuda@ufl.edu)
University of Florida IFAS Department of Entomology and Nematology
Box 110620, Gainesville FL 32611

Eric Dibble (edibble@cfr.msstate.edu)
Mississippi State University Department of Wildlife and Fisheries (emeritus)
Box 9690, Mississippi State MS 39762

Jason Ferrell (jferrell@ufl.edu)
University of Florida IFAS Center for Aquatic and Invasive Plants
7922 NW 71st Street, Gainesville FL 32653

Kurt Getsinger (Kurt.D.Getsinger@usace.army.mil)
US Army Engineer Research and Development Center
3909 Halls Ferry Road, Vicksburg MS 39180

Tyler Geer (tgeer10@gmail.com)
Clemson University Department of Forestry and Natural Resources
PO Box 340317, Clemson SC 29634

Lyn Gettys (lgettys@ufl.edu)
University of Florida IFAS Fort Lauderdale Research and Education Center
3205 College Avenue, Davie FL 33314

William Haller (whaller@ufl.edu)
University of Florida IFAS Center for Aquatic and Invasive Plants (emeritus)
7922 NW 71st Street, Gainesville FL 32653

Mark Hoyer (mvhoyer@ufl.edu)
University of Florida IFAS Fisheries and Aquatic Sciences
7922 NW 71st Street, Gainesville FL 32653

Robert Johnson (rlj5@cornell.edu)
Cornell University (retired); Racine-Johnson Aquatic Ecologists
1185 Ellis Hollow Road, Ithaca NY 14850

Contributors

Scott Kishbaugh (sakishba@gw.dec.state.ny.us)
New York State Department of Environmental Conservation (retired)
625 Broadway, 4th Floor, Albany NY 12233

Tyler Koschnick (tylerk@sepro.com)
SePRO Corporation
11550 North Meridian Street, Suite 600, Carmel IN 46032

Carlton Layne (clayne@aquatics.org)
US Environmental Protection Agency (retired); Executive Director of AERF
3272 Sherman Ridge Drive, Marietta GA 30064

John Madsen (jmadsen@ucdavis.edu)
USDA Agricultural Research Service ISPHRU
University of California-Davis Plant Sciences
MS4, 1 Shield Avenue, Davis CA 95616

Ian Markovich (ijmarkovich@ufl.edu)
University of Florida IFAS Fort Lauderdale Research and Education Center
3205 College Avenue, Davie FL 33314

Bernalyn McGaughey (bmcgaughey@complianceservices.com)
Compliance Services International
7501 Bridgeport Way West, Lakewood WA 98499

Catherine McGlynn (Catherine.McGlynn@dec.ny.gov)
New York State Department of Environmental Conservation
625 Broadway, 4th Floor, Albany NY 12233

Linda Nelson (Linda.S.Nelson@usace.army.mil)
US Army Engineer Research and Development Center (retired)
3909 Halls Ferry Road, Vicksburg MS 39180

Toni Pennington (Tpennington@esassoc.com)
Environmental Science Associates
2863 NW Crossing Drive, Suite 100, Bend OR 97703

David Petty (dpetty@ndrsite.com)
NDR Research
710 Hanna Street, Plainfield IN 46168

Megan Lee Pietruszewski (mpietru@clemson.edu)
Clemson University Department of English
801 Strode Tower, Clemson SC 29634

Candice M. Prince (cprince14@ufl.edu)
University of Florida IFAS Center for Aquatic and Invasive Plants
7922 NW 71st Street, Gainesville FL 32653

Contributors

Rob Richardson (rob_richardson@ncsu.edu)
North Carolina State University
4401B Williams Hall, Campus Box 7620, Raleigh NC 27695

John Rodgers (jrodgers@nctv.com)
Clemson University Department of Forestry and Natural Resources (emeritus)
PO Box 340317, Clemson SC 29634

Jeffrey Schardt (jeff.schardt@gmail.com)
Florida Fish and Wildlife Conservation Commission (retired)
810 Remington Avenue, Thomasville GA 31792

Patricia Shaw-Allen (pat.shaw-allen@noaa.gov)
NOAA Fisheries Office of Protected Resources
1315 East-West Highway, 13th Floor, Silver Spring, MD 20910

Joseph Sigmon (jsigmon@ufl.edu)
University of Florida IFAS Fort Lauderdale Research and Education Center
3205 College Avenue, Davie FL 33314

Donald Stubbs (donald271@verizon.net)
US Environmental Protection Agency (retired)
2301 Home Farm Court, Gambrils MD 21054

Ryan A. Thum (ryan.thum@montana.edu)
Montana State University Plant Sciences and Plant Pathology Department
119 Plant Biosciences Building, Bozeman MT 59717

Ryan Wersal (ryan.wersal@mnsu.edu)
Minnesota State University Mankato
S-143 Trafton Science Center South, Mankato MN 56001

Jack Whetstone (jwhtstn@clemson.edu)
Clemson University Baruch Institute of Coastal Ecology and Forest Science (emeritus)
PO Box 596, Georgetown SC 29442

Ben Willis (ben.willis@sepro.com)
SePRO Corporation Research and Technology Campus
16013 Watsonseed Farm Road, Whitakers NC 27891

Thomas Woolf (Thomas.woolf@mt.gov)
Montana Fish Wildlife and Parks, Aquatic Invasive Species Bureau
1420 East 6th Avenue, PO Box 200701, Helena MT 59620

Table of Contents

1.1 Impact of Invasive Aquatic Plants on Aquatic Biology

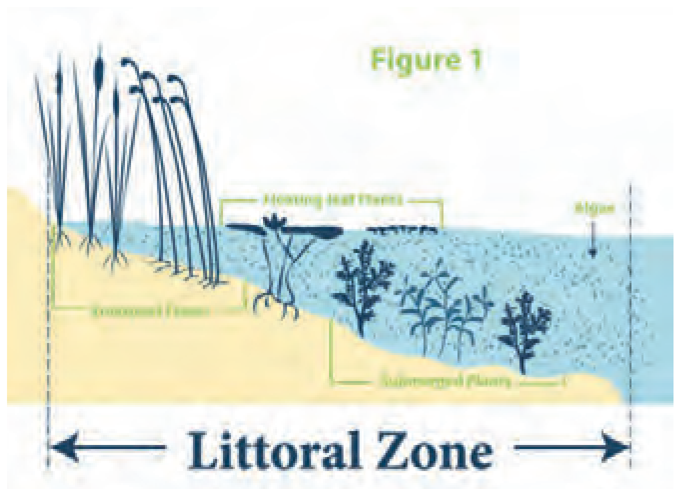
John D. Madsen: USDA Agricultural Research Service, Davis CA; jmadsen@ucdavis.edu

Introduction

Aquatic plants play an important role in aquatic systems worldwide because they provide food and habitat to fish, wildlife and aquatic organisms. Plants stabilize sediments, improve water clarity and add diversity to the shallow areas of lakes. Unfortunately, nonnative plants that are introduced to new habitats often become a nuisance by hindering human uses of water and threatening the structure and function of diverse native aquatic ecosystems. Significant resources are often expended to manage infestations of aquatic weeds because unchecked growth of these invasive species often interferes with use of the water, increases the risk of flooding and results in conditions that threaten public health.

Types of aquatic plants

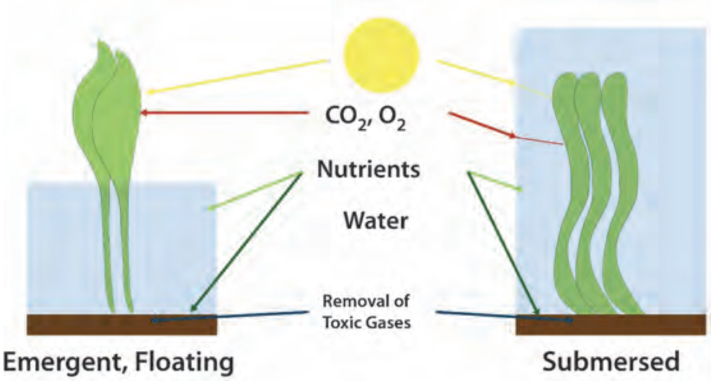
Aquatic plants grow partially or completely in water. Macrophytic plants are large enough to be seen with the naked eye (as compared to phytoplankton, which are tiny and can only be identified with a microscope) and are found in the shallow zones of lakes or rivers. This shallow zone is called the littoral zone and is the area where sufficient light penetrates to the bottom to support the growth of plants. Plants that grow in littoral zones are divided into three groups. Emergent plants inhabit the shallowest water and are rooted in the sediment with their leaves extending above the water's surface. Representative species of emergent plants include bulrush (*Scirpus* sp., *Schoenoplectus* sp.), cattail (*Typha* sp.) and arrowhead (*Sagittaria* sp.). Floating-leaved plants grow at intermediate depths. Some floating-leaved species are rooted in the sediment, but others are free-floating with roots that hang unanchored in the water column. The leaves of floating-leaved plants float more or less flat on the surface of the water. Waterlily (*Nymphaea* sp.) and spatterdock (*Nuphar advena*) are floating-leaved species, whereas waterhyacinth (Section 2.11) and waterlettuce (Section 2.12) are free-floating plants. Submersed plants are rooted in the sediment and inhabit the deepest fringe of the littoral zone where light penetration is sufficient to support growth of plants. Submersed plants grow up through the water column and the growth of most submersed species occurs entirely within the water column, with no plant parts emerging from the water. Submersed species include invasive plants such as hydrilla (Section 2.2), Eurasian watermilfoil (Section 2.3) curlyleaf pondweed (Section 2.4) and egeria (Section 2.5) along with native plants such as vallisneria (*Vallisneria americana*), coontail (*Ceratophyllum demersum*) and others.



Algae (Section 2.18) also grow in lakes and provide the basis of the aquatic food chain. The smallest algae are called phytoplankton and are microscopic cells that grow suspended in the water column throughout the lake. Dense growth of phytoplankton may make water appear green, but even the "cleanest" lake, with no green coloration, has phytoplankton suspended in the water. Filamentous algae grow as chains of cells and may form large strings or mats. Some filamentous algae are free-floating and grow suspended in the water column, but other species grow attached to plants or the bottom of the lake. Macroscopic or macrophytic algae are large green organisms that look like submersed plants, but are actually algae (Section 2.15).

What aquatic plants need

Plants have simple needs in order to grow and thrive – they require carbon dioxide, oxygen, nutrients, water and light. Plants use light energy, water and carbon dioxide to synthesize carbohydrates and release oxygen into the environment during photosynthesis. Animals use both the carbohydrates and oxygen produced by plants during photosynthesis to survive, so without plants there would be no animal life. The nutrients required in the greatest quantity by plants are nitrogen and phosphorus, but a dozen or more other minerals are also needed to support plant growth. Plant cells use oxygen in the process of respiration just like animal cells, but this is often forgotten since plants produce more oxygen than they need for their own use.



Aquatic plants inhabit an environment very favorable in one respect – most terrestrial plants must find sufficient water to survive. Aquatic plants are literally bathed in water, one of the primary requirements for plant growth. Since aquatic sediments are typically high in nitrogen and phosphorus, life might appear idyllic for aquatic plants. Once the leaves of emergent and floating–leaved plants rise above the water surface, they have a ready supply of carbon dioxide, oxygen and light. In addition, the leaves may act as a conduit for the ready disposal of toxic gases like methane and sulfur dioxide that are produced in the sediments surrounding plant roots. Given these factors, it is no surprise that emergent plants in fertile marshes are among the most productive ecosystems in the world.

Alas, life is not as easy for submersed plants. While submersed plants have easy access to the same pool of nutrients from the water and the sediment, the availability of light and carbon dioxide is significantly reduced since most submersed plants live completely under the water. Light must penetrate through the water column to reach submersed plants; therefore, much less light energy is available to them. Also, carbon dioxide must be extracted from the water, an environment in which carbon dioxide is present in much lower concentrations and diffuses much more slowly than in air. As a result, submersed plants are much less productive and produce less biomass than emergent and floating plants and the primary factors limiting their growth are the availability of light and carbon dioxide. Some highly productive plants have developed the means to increase their access to light and carbon dioxide. For example, species such as hydrilla form dense floating mats or canopies on the surface of the water, which allows them to capture light energy that is less available near the bottom of the water column. These productive (and often invasive) aquatic plants form dense colonies that interfere with human uses of the littoral areas, increase flooding risk and shade out other plants – including most native species – that do not form canopies.

Lake ecology

Trophic state

Trophic state describes the overall productivity (amount of plants or algae) of a lake, which has implications for the biological, chemical and physical conditions of the lake. For example, aquatic animals use plants as a food source, so unproductive lakes do not support large populations of zooplankton, invertebrates, fish, birds, snakes and other animals. The trophic state of a lake is directly tied to the overall algal productivity of the lake and ranges from very unproductive to highly productive. Because phytoplankton typically control lake productivity, factors that increase algal productivity also increase the trophic state of the lake. Algal biomass in a lake is estimated by measuring the concentration of chlorophyll in the water; hence, lake chlorophyll concentration is a direct measure of lake trophic state.

Chlorophyll is directly related to phosphorus concentration in the lake, so phosphorus is also considered a direct measure of lake trophic state. Lake transparency is the most widely measured characteristic to determine trophic state because growth of algae increases water turbidity – high algal growth reduces water clarity, which suggests high productivity. Trophic state can be measured with a Secchi disk because most turbidity in lakes is caused by suspended algae. Since increased algal growth makes the water less transparent, Secchi disk depth is a measure of lake trophic state. Chlorophyll, phosphorus and Secchi disk depth are measured in different units. The Trophic State Index (TSI) employs equations that allow users to develop a single uniform number for trophic state based on any one of the three factors alone or on the average of all three factors (chlorophyll, total phosphorus or Secchi disk depth). This tool is useful to compare trophic state data collected by differing methods and has empowered hundreds of lay monitors to collect trophic state data using only a Secchi disk to estimate water clarity.

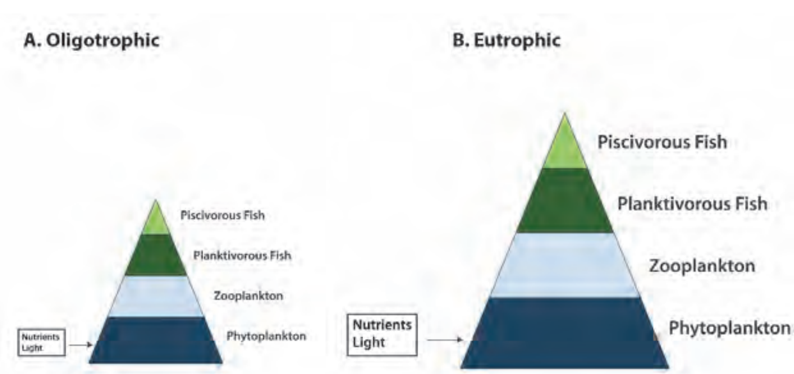
Four terms are commonly used to describe lake trophic state. Oligotrophic lakes are unproductive with low nutrients (phosphorus < 15 μg/L) and low algal productivity (chlorophyll < 3 μg/L). Transparency, as measured by the Secchi disk method, is greater than 13 feet. Oligotrophic lakes are typically well-oxygenated and often support cold-water fisheries in the northern US. Mesotrophic lakes are moderately productive, with intermediate levels of chlorophyll, nutrients and water clarity. Mesotrophic lakes may support abundant populations of rooted aquatic plants and often have cool-water fisheries. Eutrophic lakes are highly productive, with high levels of phosphorus and chlorophyll. Water clarity is low and generally ranges from 3 to 8 feet as measured by the Secchi disk method. Eutrophic lakes may support bass fisheries but rarely have productive open-water fisheries. Hypereutrophic lakes have very high phosphorus and chlorophyll levels and water clarity is usually less than 3 feet. In most cases, hypereutrophic lakes are the result of nutrient loading from human activity in the watershed. Algal growth dominates in the lake and few or no rooted submersed plants are present.

Trophic state	Chlorophyll concentration (μg/L)	Total phosphorus concentration (μg/L)	Water clarity (by Secchi disk, in feet)	Trophic State Index	Description
Oligotrophic	< 3	< 15	> 13	< 30	Very low productivity Clear water Well oxygenated Few plants and animals
Mesotrophic	3-7	15-25	8-13	40-50	Low to medium productivity Moderately clear water Abundant plant growth
Eutrophic	7-40	25-100	3-8	50-60	Medium to high productivity Fair water clarity Dense plant growth
Hypereutrophic	> 40	> 100	< 3	> 70	Very high productivity Poor water clarity Limited submersed plant growth, algae dominate

Studies of sediment cores from across the US have verified that many lakes were naturally mesotrophic or eutrophic before Europeans settled in the US, which conflicts with the assumption that all "pristine" lakes are oligotrophic. The nutrient status or trophic state of lakes that are unaffected by human activity is a function of the watershed and its geology. That being said, human activity that causes nutrient runoff into lakes can shift a lake to a higher trophic state, which alters many biological and chemical attributes of the lake. There are many examples of pollution-degraded lakes, but the water quality of many lakes has improved since the passage of the first Clean Water Act in 1972 and these lakes are returning to their historic water quality levels due to efforts to restore our waterways.

Productivity in lakes

As mentioned above, algae and macrophytic plants are the basis for lake productivity. Plants take up nutrients, water and carbon dioxide from the environment and use light energy to produce carbohydrates and sugars, with oxygen as a byproduct. Herbivores such as crustaceans and insects consume aquatic plants and use energy from the plants to grow. Forage fish such as minnows and bluegill consume these herbivores and use energy from the herbivores to grow. Fish-eating fish such as trout, bass, pike and walleye



eat these forage fish and use energy from the forage species to grow (Section 1.2). Because each level of this feeding system is based on the energy of the level below it, this system is often described as a food pyramid. Oligotrophic lakes with few nutrients and little plant production have small pyramids, whereas eutrophic lakes with much higher nutrient concentrations, more total plant growth (algae and rooted plants) and more fish have larger pyramids. This relationship has been recognized by the aquaculture industry and fertilizer is frequently added to production ponds to increase fisheries productivity. However, changes in water quality can increase populations of undesirable fish along with populations of more desirable species in reservoirs and in natural systems.

Food chains in lakes

A food chain is a depiction of what various organisms in an ecosystem consume. Food chains begin with algae and plants, which are followed by herbivores, small forage fish and finally by the top-level predator. There may be a hundred species in a lake, so the food chain is often simplified to include only the dominant species. Phytoplankton form the base of the food chain in a typical pelagic (open-water) zone. Phytoplankton are consumed by zooplankton (small crustaceans) that are suspended in the water. Zooplankton are in turn eaten by smaller fish such as yellow perch. Yellow perch are then consumed by the top predator such as walleye.

The food chain in the littoral zone is different. Some algae are present – both as phytoplankton and as algae growing on plant surfaces – but much of the food is derived from macrophytic plants. Most macrophytes are consumed only after they have died and partially decomposed into detritus. Detritus is eaten primarily by aquatic insects, invertebrates and larger crustaceans. These detritivores, which live on or near the lake bottom, are in turn consumed by the dominant littoral forage fish such as bluegill sunfish. Lastly, forage fish are consumed by the top predator such as largemouth bass.

Littoral and cold-water pelagic zone food chains are often isolated from each other and almost function as two separate ecosystems within the same lake. The substantial changes caused by shifts between these food chains are exemplified by the history of Lake St. Clair in Michigan. Lake St. Clair only looks small compared to the Great Lakes it lays between – Lakes Huron and Erie. In fact, it is a 430 square mile lake with a maximum depth of 30 feet, although over 90% of the lake is 12 feet deep or less. This shallow lake was very turbid before 1970, with a Secchi disk transparency of only 4 feet. Rooted plants grew in about 20% of the lake and Lake St. Clair was home to a world-class commercial and recreational open-water walleye and yellow perch fishery. Lake St. Clair was invaded in the 1980s by the zebra mussel, an invasive bivalve (clam) that filters water by consuming suspended phytoplankton and the nutrients associated with them. Zebra mussels filtered the water of Lake St. Clair so effectively that water transparency more than doubled a few years after their invasion. Rooted plants expanded to almost 80% of the lake due to increased light penetration and the fishery completely changed. Walleye and yellow perch can still be found, but the former open-water fishery is now used largely for recreational angling for largemouth bass, a typical littoral zone predator.

Aquatic plant communities

Native aquatic plant species tend to separate into depth zone bands (referred to as depth zonation), with a mix of species found in each depth zone. Submersed plants may be found in water as deep as 30 feet or more in oligotrophic lakes and distinct bands of vegetation are visible to the shoreline. Plants in oligotrophic lakes are adapted to low levels of nutrients and carbon dioxide. Light penetrates easily to 30 feet or more and light levels are not limiting, but plants are typically very short. Submersed aquatic mosses also grow at water depths of up to 200 feet in Crater Lake in Oregon. Plant diversity is often relatively low and native plants in oligotrophic lakes rarely form populations that are substantial enough to cause problems.

Depth zonation in mesotrophic lakes is likewise pronounced, with submersed plants growing in water as deep as 15 to 20 feet. Submersed plants may grow to reach the surface of the water, but this growth is typically localized and occurs in water that is less than 10 feet deep. Plant species diversity is usually at a maximum in mesotrophic lakes; numerous plant growth forms are present and result in a multi-layered plant canopy. Light penetration may limit plant growth but plants grow at depths greater than in eutrophic lakes and the total amount of plant growth in mesotrophic lakes is often as high as in eutrophic systems. Nutrients rarely limit plant growth in mesotrophic systems and growth of aquatic species is almost completely dependent on light penetration. Residents living next to reservoirs and lakes often report changes in plant coverage from year to year; these changes are typical of dynamic mesotrophic systems and are usually the result of changes in light penetration.

Depth zonation in eutrophic lakes is much less pronounced, with plant growth typically occurring at maximum depths of only 12 to 15 feet. Plant abundance is high, but plant diversity is much lower than in mesotrophic lakes and erect and canopy-forming plants predominate because light is often limited due to growth of phytoplankton. Native plants often produce populations that are large enough to be nuisances, particularly in high-use areas such as boat ramps and swimming areas. Light strongly limits plant growth and canopy-forming plants have a distinct advantage over plants that do not form canopies.

Hypereutrophic lakes typically have poorly developed aquatic plant communities and plants rarely grow in water more than 6 feet deep. Some emergent and floating plants can be found, but submersed plant growth is greatly reduced and typically only canopy-forming species are able to establish. Plants that are able to colonize hypereutrophic lakes often grow to nuisance levels. High algal production results in dense blooms that intercept available light. As a result, plant diversity is low and the abundance of rooted plants is typically lower than in eutrophic lakes.

So what should a typical lake look like? Well, that depends. Without human-mediated nutrient loading from sewage treatment plants and runoff from fields and residential areas, hypereutrophic lakes would be rare occurrences. Therefore, the natural state of a typical lake would include a littoral zone dominated by aquatic plants. Even in eutrophic lakes, nuisance populations of native plants would likely be localized and would cause problems only when the plants interfere with recreational or other uses. However, the introduction of invasive exotic plants changes this dynamic, even in oligotrophic lakes.

Invasive plants

Invasive aquatic plants are generally defined as nonnative (from another geographic region, usually another continent) plant species that cause ecological and/or economic harm to a natural or managed ecosystem. Invasive aquatic plants often cause both economic and ecological harm.

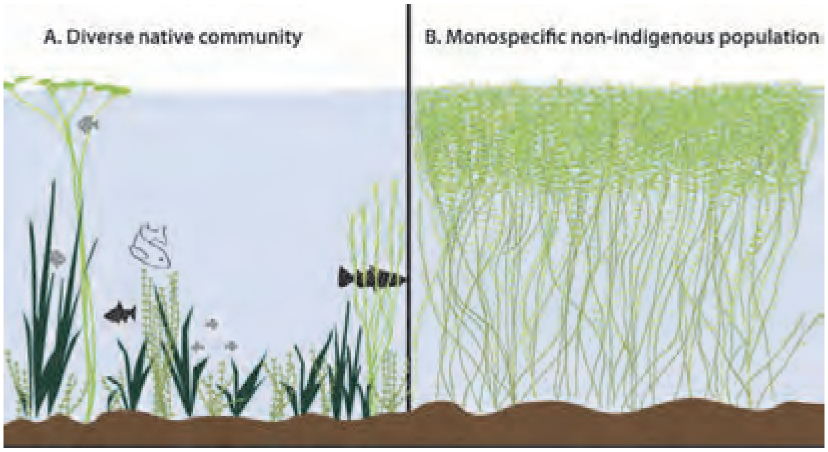
As invasive plants expand in a new area, they suppress the growth of native plants and cause localized extinction of native species. For instance, when Eurasian watermilfoil invaded Lake George in New York, growth of this exotic species reduced the total number of species in a permanent research plot from 21 to 9 over a three-year period. Invasive plant species can invade a particular zone of the depth profile and suppress the native plant species that normally inhabit that area. Colonization by invasive species may be less damaging in oligotrophic lakes because native plants can grow at much greater depths than invasive species. Native plants often persist in areas of mesotrophic lakes that are shallower and deeper than those colonized by invasive plants. Invasive plants dominate to the borders of eutrophic and hypereutrophic lakes, with native plants often confined to a shallow fringe around the lake.

Economic impacts	Ecological effects
Impair commercial navigation	Degrade water quality
Disrupt hydropower generation	Reduce species diversity
Increase flood frequency, duration and intensity	Suppress desirable native plants
Impair drinking water (taste and odor)	Increase extinction rate of rare, threatened and endangered species
Habitat for insect-borne disease vectors	Alter animal community interactions
Recreational navigation impairment	Increase detritus buildup
Interfere with safe swimming	Change sediment chemistry
Interfere with fishing	
Reduce property value	
Endanger human health, increase drowning risk	

Summary

Invasive plants reduce native plant growth and impede human uses of waters by forming dense surface canopies that shade out lower-growing native plants and interfere with water flow, boat traffic and fishing. Dense surface canopies also radically change the habitat quality for fish. Dense plant beds provide a place for small forage fish to hide and reduce the ability of predatory fish such as bass and northern pike to see their prey. This tends to lead to a large number of small, stunted forage fishes and poor production of game fishes.

Invasive plants also reduce water quality. While the increased biomass and dense canopies formed by invasive species tend to increase water clarity, they also lead to increased organic sedimentation. The fate of all lakes over geological time is to progress from lakes to wetlands to marshes to upland areas as lakes fill with sediments due to erosion and accumulation of organic matter. Exotic plants are also significantly more productive than native species and increase the rate of nutrient loading in the system by utilizing nitrogen and phosphorus from the sediment. For example, curlyleaf pondweed has been implicated in increased internal nutrient loading in Midwestern lakes because the plants absorb nutrients from the sediments and grow throughout the spring and summer, then die and release the nutrients into the water. Water also becomes stagnant under dense plant canopies that suppress or prevent oxygen recirculation. In addition, the amount of dissolved oxygen under dense plant canopies may be insufficient to support desirable fish species and may result in fish kills.



Many animal species are linked to specific native plant communities and the diversity of native communities provides a variety of habitats for aquatic insects and other fauna. Invasive plants reduce the diversity of native plant communities, which leads to a reduction in the diversity of both fish and aquatic insects. Therefore, invasive plants are harmful to the diversity and function of aquatic ecosystems and can have significant adverse impacts on water resources.

Photo and illustration credits:

Page 1: Littoral zone; Minnesota Department of Natural Resources
Page 2 upper: Aquatic plants illustration; John Madsen, USDA ARS
Page 2 lower: Secchi disk; Margaret Glenn, University of Florida
Page 3: Food pyramids; John Madsen, USDA ARS
Page 6: Heterogeneous and homogeneous plant communities; Robert Doyle, Baylor University

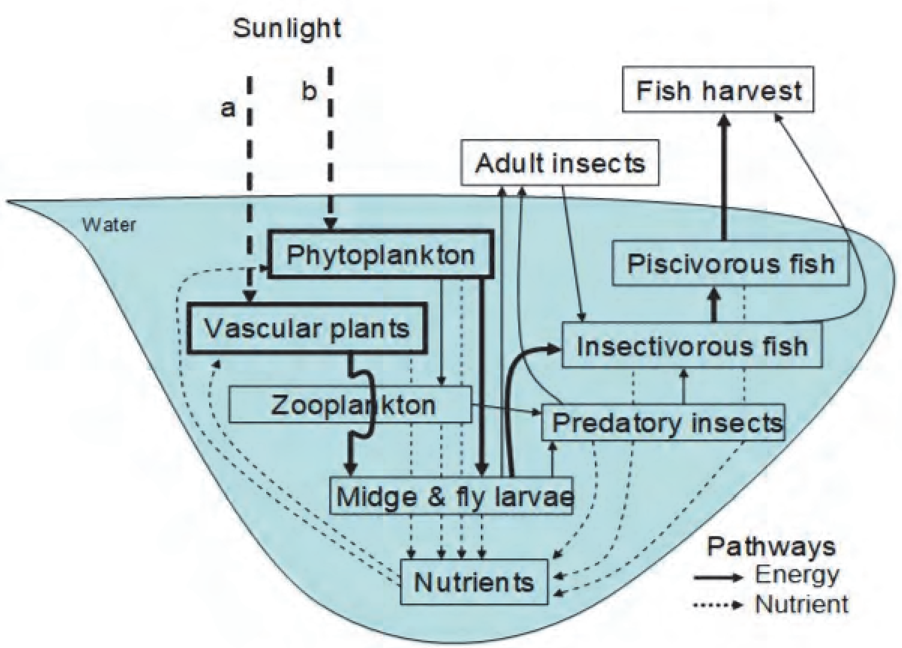
1.2 Impact of Invasive Aquatic Plants on Fish

Eric Dibble: Mississippi State University, Mississippi State MS (emeritus); e.dibble@msstate.edu

Introduction

Many species of fish rely on aquatic plants at some point during their lives and often move to different habitats based on their growth stage. Young fish use the cover provided by aquatic vegetation to hide from predators and their diets may be dependent on algae (Section 2.18) and the microfauna (e.g., zooplankton, insects and larvae) that live on aquatic plants. Mature fish of some species move to more open waters to reduce foraging competition and also include other fish in their diets. Also, different fish prefer different types of habitats and will move to a new area if foraging conditions in their preferred location decline due to excessive growth of aquatic weeds.

The energy cycle



The energy that supports all life on earth – including life in lakes – originates from sunlight. Vascular plants and phytoplankton (algae) capture light in the chloroplasts of their cells and convert it to energy through photosynthesis (Section 1.1). Aquatic plants and phytoplankton use this energy to subsidize new growth, which is consumed and used as an energy source by aquatic fauna. For example, phytoplankton is eaten by zooplankton, or vascular plant tissue is eaten by insect larvae. The zooplankton and insect larvae are then eaten by larger insects and/or insect-eating fish. This energy cycle continues with ever-larger organisms consuming smaller ones and provides a vivid illustration of the "trickle-up economics" of energy cycling. As this example demonstrates, the vegetated aquatic habitat that is essential for insects and small fish can be a critical component in the process that fosters growth of harvestable fish.

The relationship between fish and aquatic plants

The abundance of some fish declines with increased plant densities. For example, populations of white bass (*Morone chrysops*), gizzard shad (*Dorosomoa cepedianum*) and inland silverside (*Menidia beryllina*) generally decline where heavy vegetation is present. In contrast, many juvenile and some adult fish prefer habitats with aquatic vegetation; in fact, over 120 different species representing 19 fish families have been collected in aquatic plant beds. Sites with vegetation generally have higher numbers of fish compared to non-vegetated areas. In fact, densities of greater than 1 million fish per acre have been reported in areas containing a diversity of aquatic plants. Very few of these fish, however, survive to become large adults, so high numbers of small fish do not always result in populations of large mature fish. Excessive growth of aquatic plants promotes high populations of small fish in contrast to more diverse and balanced plant populations. Reduced plant densities due to weed management activities, boat traffic and/or natural senescence may change or cause the loss of invertebrate food sources. However, studies of lakes where invasive plants were treated with early applications of herbicides to allow native plants to reestablish have revealed that removal of exotic weeds has little impact on invertebrate populations and no measurable effect on fish communities.

Fish and their affinity for plants

Fish	Plant affinity	Life stage			Relationship		
		Larvae	Juvenile	Adult	Spawn	Forage	Predator avoidance
Bluegill sunfish	High	X	X	X	X	X	X
Common carp	High	X	X	X	X	X	X
Largemouth bass	High	X	X	X	X	X	X
Muskie	High	X	X	X	X	X	X
Northern pike	High	X	X	X	X	X	X
Black crappie	Moderate		X	X	X	X	X
Smallmouth bass	Moderate		X	X		X	X
Yellow perch	Moderate	X	X			X	X
White crappie	Low		X			X	
Salmon, trout	Low		X				X
Shad	Low	X					
Walleye	Low			X		X	

Bluegill sunfish (*Lepomis macrochirus*) are often referred to as the "kings" of plant-loving fishes and strongly prefer vegetated habitats throughout much of their lives. There are many different types of small sunfishes, but the bluegill is likely one of the most popular freshwater fish in North America. The bluegill is the most intensely studied freshwater fish in the US and is considered to be a "lab rat" by fish biologists. In addition to its popularity with scientists, the bluegill has been widely stocked, carefully managed and regularly harvested in natural and artificial systems throughout the US. Bluegill is a premier food fish and is called "pan fish" in the North and "bream" in the South.

Similar to other sunfishes, bluegill often move to new habitats as they age. Bluegill sunfish spawn and nest in colonies near areas of submersed vegetation, where soft sediment and plants are cleared. Bluegill larvae are transparent and can safely move from shallow shoreline habitats to open water where they feed on plankton. As the larvae grow larger and develop color, they become more attractive to predators and seek refuge among aquatic vegetation where they feed on insects, midges and small crustaceans. Juveniles and small adult fish remain among shoreline plants and feed on the food they can capture; as they grow, they may shift to feeding on larger crustaceans, insects and amphipods. As fish mature, grow larger and change color, their chances of being eaten by predators decrease and they shift to more optimal feeding grounds. Bluegill continue to feed in vegetated habitats where they can avoid larger predators until they reach approximately 8 inches in length. Fish of this size are large enough to escape most of the risk of predation, so these mature bluegill will venture away from the complex structure provided by plants and move to feed in open water. This reduces feeding competition among bluegill and provides access to larger fish that bluegill consume to supply the energy needed for continued growth. Bluegill are not considered herbivores, but they do consume plant material, most likely by accident as they forage for insects and crustaceans living on aquatic plants. Aquatic plants thus play a critical role in the growth of bluegill sunfish by hosting insects, crustaceans and invertebrates that are eaten by young fish and by providing cover that allows young fish to hide from predators.

Fish populations in lakes with a diverse assemblage of phytoplankton, aquatic plants and habitats tend to be stable. This is a general ecological principle that applies to wildlife, fish and other organisms. However, the bluegill sunfish illustrates why it is unwise to make specific “ironclad” statements regarding the habitat requirements of fish. As noted above, bluegill sunfish have very close associations with aquatic plants but can also become quite large and develop robust populations in managed fish ponds that lack aquatic plants. This apparent conflict is partially explained by the concept that bluegill food webs may be based more on phytoplankton in ponds where the predator-prey relationship has been simplified.

Largemouth bass (*Micropterus salmoides*) are stocked throughout the world and are among the world’s top freshwater game fishes. Largemouth bass are plant-loving and are closely associated with aquatic plants, spending much of their lives in or around vegetated habitats. Adult largemouth bass diligently protect their nests and offspring from predators. The structure provided by moderate densities of submersed plants improves nesting success, but an overabundance of plants can reduce nesting success. Larvae of largemouth bass feed mostly on microcrustaceans and juveniles consume larger (but still small) crustaceans, whereas mature largemouth bass primarily eat aquatic insects and small fishes (e.g., bluegill, shad and silverside). Aquatic plants serve as critical habitats that support the prey that largemouth bass rely so heavily on through their lives. These prey resources directly or indirectly influence growth and the ability of largemouth bass to overwinter and survive adverse conditions. Therefore, the abundance of largemouth bass is strongly correlated with the abundance of submersed vegetation in its habitat. However, this correlation varies based on the types and densities of the plant species in the habitat.

Smallmouth bass (*Micropterus dolomieu*) prefer deeper, cooler waters with rocks and/or woody cover and generally avoid shallow water that is dominated by aquatic plants. However, like the largemouth bass, young smallmouth bass prey on the insects, crustaceans and other microfauna that are hosted by aquatic plants. More mature smallmouth bass consume crayfish, larger insects and other fishes (including shad). Shad feed primarily on phytoplankton and detritus and avoid aquatic vegetation, so the diet of adult smallmouth and largemouth bass may be dependent on prey fish that do not prefer a vegetated habitat, especially in reservoirs. Smallmouth bass protect their nests and offspring but are less selective of nesting location and will choose nesting sites in shallow water if the water has some form of cover. This cover may be provided by aquatic plants, but most sites have cover in the form of rocky outcrops or overhanging woody debris. Because young smallmouth bass consume microfauna associated with aquatic plants and sometimes use aquatic plants to avoid predators, their relationship with aquatic plants is moderate.

White crappie (*Pomoxis annularis*) have a low affinity for aquatic plants since they typically spawn in nests away from vegetation and spend much of their time as adults and juveniles in open water. However, aquatic plants can directly affect spawning and indirectly influence the diet available to young white crappie. Research suggests that excessive amounts of aquatic plants may reduce spawning success of a nesting colony of white crappie. In addition, the presence of aquatic plants may deter nesting altogether. Eggs of white crappie have been found in aquatic vegetation; however, this is most likely incidental drift of eggs from nearby nesting sites. Larval white crappie feed primarily on microfauna, whereas juveniles feed on insect adults and larvae (i.e., midges and water boatmen) that frequently inhabit vegetated habitats.

Black crappie (*Pomoxis nigromaculatus*) are more closely associated with aquatic plants than their cousins, the white crappie, and have a moderate affinity for plants. Adult black crappie prefer sites with plants – including submersed, emergent, flooded and even inundated terrestrial species – for nesting and spawning and are more likely than white crappie to care for nests and offspring. Like white crappie, they also rely on many of the insects that live in aquatic vegetation. In fact, young black crappie rely heavily on insect larvae and other microfauna that are strongly associated with vegetated habitats.

Gizzard shad (*Dorosoma cepedianum*) are small fish that are widely distributed and are frequently stocked in reservoirs as prey for fish-eating fishes such as crappie and striped, largemouth and other bass. Gizzard shad are not usually considered to be associated with aquatic plants; as larvae, they may rely on food resources from vegetated habitats but their affinity for these habitats is low. Larvae of gizzard shad feed on algae, protozoans and microfauna, whereas adults are more herbivorous and consume phytoplankton in the water column and detritus (decomposed vascular plants) in the sediment. Gizzard shad usually spawn at or near the surface of the water and broadcast their eggs. Eggs drift on the water and can attach to any surface, but it is not uncommon to find egg masses attached to aquatic vegetation. In fact, some egg masses are so large that stems of emergent aquatic plants may collapse under their weight.

Common carp (*Cyprinus carpio*) are invasive, exotic, nuisance species that are detrimental to many aquatic systems. Common carp are frequently found in reservoirs and natural lakes and are associated with shallow areas that have soft sediments and abundant submersed vegetation. Common carp are omnivorous bottom feeders whose diets are composed primarily of organic detritus (mostly in the form of dead plant material) and benthic organisms, including insect adults and larvae, crustaceans, snails, clams and almost anything else organic that they encounter. The mouth parts of common carp are specialized for foraging for hard items (i.e., plants and animals) within soft sediments and among the roots of aquatic plants. Adult fish typically spawn in shallow water inhabited by aquatic plants, where plant stems and leaves serve as attachment sites for fertilized eggs after spawning. Eggs require oxygen to survive; egg attachment to plant structures prevents eggs from settling into soft sediments that lack the oxygen needed for egg survival.

Salmon and **trout** are not usually associated with aquatic plants and their affinity for vegetated habitats is typically thought to be low. However, some trout species may develop indirect relationships to aquatic plant habitats after the fish are introduced into cool reservoirs and natural lakes. For example, the diet of trout in these systems is often dominated by adults, nymphs and larvae of caddisfly, stonefly, cranefly and mayfly, all insects that are frequently associated with aquatic vegetation. This observation, along with reports that navigation and migration of adult salmon and trout may be hindered by dense beds of invasive aquatic plants, suggests that the relationship of salmon and trout to aquatic vegetation may be complex.

Aquatic plants play an important role in the foraging and reproductive strategies of **northern pike** (*Esox lucius*), which typically avoid strong currents and have strong affinities for dense beds of aquatic plants during feeding and spawning. Northern pike primarily feed on other fish by using "ambush" foraging strategies—they wait and strike at prey with a burst of swimming energy. Northern pike are among the first fish to spawn in early spring and broadcast their adhesive-coated eggs on shallow weedy areas. After being released, the eggs drift and settle on submersed vegetation, where they attach and are well-oxygenated.

Muskellunge or **muskie** (*Esox masquinongy*) are rarely found far from aquatic plants during any stage of their life. They rely heavily on prey resources (i.e., fish, young ducks, frogs and muskrats) that live in vegetated habitats. Muskie spawn later than northern pike, but utilize similar spawning tactics and rely on plants to successfully reproduce. Eggs of muskie also have an adhesive coating and adhere to plant structures after being broadcast.

Walleye (*Stizostedion vitreum*) are not classified as having a strong affinity for aquatic vegetation, despite reports that walleye are sometimes caught near vegetation. However, vegetation in flooded marshes can provide a substrate for spawning, and populations of some species used by walleye as prey (e.g., yellow perch) do rely on vegetated habitats. Walleye are not tolerant of increases in turbidity or suspended sediment. Therefore, aquatic plants may play an indirect role in improving the walleye habitat in some systems by filtering sediments and decreasing water turbidity.

Adults of **yellow perch** (*Perca flavescens*) are typically found in open waters with moderate levels of aquatic plants, but when young their affinity for plants is relatively high. Yellow perch are frequently associated with rooted aquatic vegetation. Successful spawning sites typically contain some form of structure, most often in the form of submerged aquatic plants. Like bluegill, young yellow perch switch habitats as they mature. As clear larvae, they feed in open water on zooplankton; once they become pigmented, they return to shallow water with vegetation where they feed on small fishes and insects along the bottom.

Plants provide critical structure to aquatic habitats

The shade created by leafy plants is important to many visual feeders because shade can improve visibility for both selecting prey and avoiding predators. Vegetated aquatic habitats also provide food for young and small fish of many species while protecting them from predators. The abundance and diversity of aquatic fauna eaten by small fish are higher in vegetated habitats than in areas with no plants because leaves and stems provide a surface for attachment; also, small gaps among plants can provide a place for fauna to escape and hide from predators. As vegetated habitats become more complex, the risk of small fish becoming prey may be decreased. However, the ability of fish to forage declines as vegetated habitats become more complex as well. Visual barriers created by leaves and stems may make it more difficult for fish to find and capture prey, whereas swimming barriers that result from dense vegetation can increase search time by reducing maneuverability and swimming velocity. For example, the rate at which sunfish successfully capture prey declines with an increase in structurally complex vegetated habitats. Some fish have developed tactics to address the negative aspects (i.e., reduced food availability accompanied by increased efforts to capture prey) associated with densely vegetated areas. The largemouth bass, for example, changes foraging tactics in complex habitats and switches from actively pursuing prey to ambushing them as they drift or swim by.

Plants influence growth of fish

Studies have shown that aquatic plant abundance affects the growth and health of fish, especially plant-loving fish such as the sunfishes. Habitats with moderate amounts of aquatic vegetation provide the optimal environment for many fish and enhance fish diversity, feeding, growth and reproduction. Conversely, both limited and excessive plant growth may decrease fish growth rates.

High densities of plants can reduce the growth and health of largemouth bass and of black and white crappie, most likely by reducing foraging efficiency. Fisheries scientists have predicted that largemouth bass growth significantly declines in systems with > 40% coverage of aquatic plants and that maintaining plant beds at an average standing crop of 5 tons of fresh weight per acre (4 ounces per square foot) would improve foraging efficiency of largemouth bass. A total removal of plant biomass exposes forage fish and can, at least temporarily, increase growth of predator fish species (i.e., largemouth bass, black and white crappie, bluegill and other sunfishes) that rely heavily on the prey that inhabits vegetated habitats.

Rapid removal of aquatic plants can alter foraging behaviors and encourage young largemouth bass to switch to eating fish sooner in life, which results in more rapid growth. Conversely, young sunfish grow most quickly in vegetated habitats because when plants are absent or sparse, competition for forage sources increases among these fish; less food resources are available to them and growth slows. However, growth of these fish can also be slowed when plant density is too high, especially in shallow-water areas where plants form monotypic beds.

Plants influence spawning

Studies suggest that the structure provided by plant beds is important to fish reproduction. In fact, many fish in North America are “obligate plant spawners” that directly or indirectly require aquatic plants in order to successfully reproduce. At least a dozen fish families use vegetation as nurseries for their young and reproductive success of nest spawners is improved when they have access to sites with aquatic vegetation and/or some form of structure. Fish can derive a number of benefits from nesting near aquatic plants. For example, vegetation can protect nest sites from wave action and sedimentation that can harm eggs and small fish. Also, parents often use aquatic plant patches or edges as “backing” to protect nests from predators. In addition, many fish that live among aquatic plants are visual feeders and the shade produced by overhanging leaves and plant canopies improves visual acuity so fish can find prey – and avoid becoming prey – with greater success. The shallow areas preferred for spawning by nesting fish are not static and can change over time so that a formerly ideal nesting site can become less than perfect. These areas can become overgrown with aquatic plants, which can hinder optimal spawning. Also, nesting fish can change the composition of the littoral zone by disturbing or altering plant growth, which could affect future nesting success.

Plants influence the physical environment

Aquatic plants can change water temperatures and available oxygen in habitats, thus indirectly influencing growth and survival of fish. The amount of oxygen a fish uses during the course of a day is referred to as daily oxygen consumption rate. High numbers of large fish are not usually found in warm-water habitats that are low in dissolved oxygen because larger fish in warmer water need more oxygen; however, smaller fish are more tolerant of such conditions. Shallow areas where aquatic plants are present and water temperatures increase quickly are inhabited by small fish more frequently than large fish because small fish have lower oxygen consumption rates and can tolerate the reduced oxygen available in these habitats. Dense monotypic beds of weeds in shallow-water habitats can negatively impact fish habitats. The structure resulting from dense growth of stems and leaves can interfere with water circulation and surface exchange of atmospheric oxygen, resulting in high water temperatures and low dissolved oxygen. These conditions can seriously impact fish health; in fact, it is not uncommon to have localized fish kills in areas with extremely dense aquatic weeds. Dense plant beds sometimes have relatively open areas that allow water circulation and oxygen exchange to occur. These areas are usually temporary, but they can serve as important refuges for fish during periods when oxygen levels are low in the rest of the weed bed. Plant beds that are managed for fish habitats should include open areas such as patches and/or lanes to improve the water circulation and oxygen exchange that are important to fish health.

The "perfect" lake: artificial and natural systems

Before determining the optimal amount and type of aquatic plants needed to create "perfect" conditions for fish growth, it is important to recognize that the two types of water systems – artificial and natural – differ from one another and present different challenges for management of aquatic plants. Both types of system can be found throughout the US; as a result, the species of fish that inhabit them (and angler goals) vary by location and contribute to management challenges. As noted above, most fish require some sort of structural habitat at some point in their lives. A diversity of structures provides a diversity of habitats, which can support many different types of aquatic organisms, including numerous species of fish. Therefore, a critical goal in managing artificial and natural water systems should be the maintenance of diverse habitats within the littoral zone, which can be accomplished by ensuring that a variety of plant species are available.

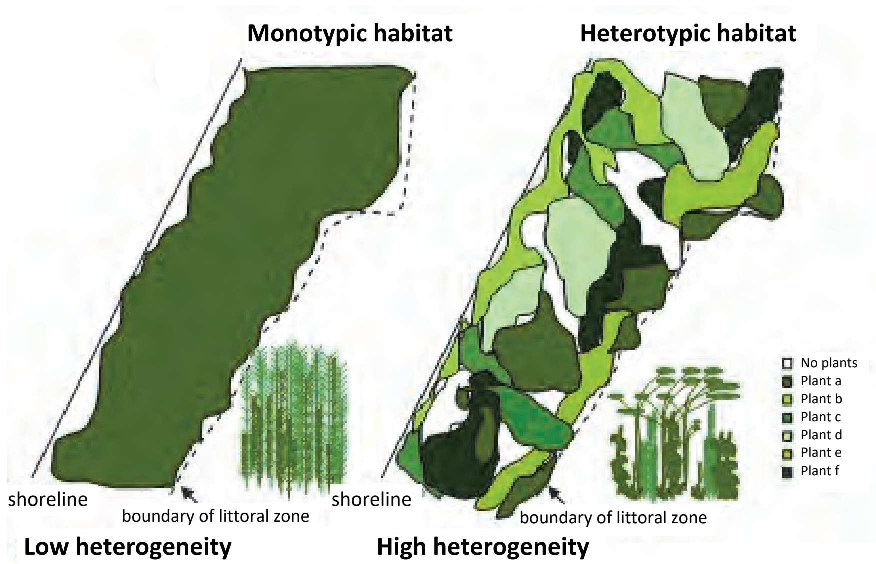
Reservoirs

Reservoirs are typically young (< 100 years old) artificial systems constructed to prevent flooding, generate electrical power and/or provide navigation for barge traffic. Much of a reservoir is an artificial basin on a flooded – but formerly terrestrial – site; therefore, few reservoirs have naturally occurring populations of native aquatic plants. The sediments of many reservoirs hold seed banks of terrestrial plants that will not germinate under flooded conditions. As a result, the sediment is often a barren benthic mud that provides ideal conditions for invasion by exotic plants. In fact, many reservoirs in the US have been taken over by invasive aquatic weeds and plant diversity is typically very low.

Fish may naturally inhabit reservoirs, but providing fish habitat is often a byproduct of the reservoir's construction and is rarely intentional. Reservoirs in the southern US are typically stocked with a variety of plant-loving fish, including largemouth bass and bluegill sunfish. As noted earlier, aquatic plants play a critical role in the growth of these fish by hosting prey such as insects, crustaceans and invertebrates and by providing cover that allows fish to hide from predators. However, dense monotypic beds of aquatic weeds can restrict the benefits associated with a vegetated habitat by reducing fish foraging ability. This results in a fish population with high numbers of small individuals that fail to grow large, a condition sometimes referred to as a "stunted population." Such populations consist of many individuals feeding in dense habitats which provide better forage resources for smaller individuals, but which restrict foraging opportunities for larger fishes. A plant density that results in coverage of 20 to 60% of the surface area within the littoral zone generally provides the best fish habitat and recreational opportunities in reservoirs.

Natural lakes

Many lakes form as a result of natural events such as flowing water, earthquakes and animal activities like dam building, but most natural lakes in the northern US are the result of glacial disturbance. These systems were formed many years ago (most recently ten thousand years ago) and are often vegetated by diverse collections of native and endemic aquatic plants. Therefore, management of natural lakes differs significantly from methods used in reservoirs, which are usually dominated by monocultures of invasive species.



Natural lakes are diverse in both aquatic plants and fish. Like reservoirs, most of the fish in natural lakes require a structural habitat at some point in their lives. In fact, many are plant-loving fish that choose to spend much of their life feeding and growing in vegetated habitats. The diversity of native and endemic aquatic plants furnishes the littoral zone with a wide variety of structures that differ in size and plant composition, a condition referred to as habitat heterogeneity. This diverse habitat is home to a number of fishes adapted to this environment, including largemouth bass, bluegill, crappie, northern pike, muskie, young perch and walleye.

Summary

Most freshwater fish rely on aquatic plants at some point during their lives and prefer specific habitats based on their growth stage. Young fish use aquatic vegetation as a food source – both by directly consuming plants (in most cases incidentally) and by foraging for the microfauna associated with the plants – and as cover to hide from predators. Mature fish move to more open waters to increase foraging success and consume other fish to supplement their diets. Nesting, growth and foraging success of plant-loving fish are influenced by plant composition and density. While many fish require some aquatic vegetation for optimal growth, excessive amounts of aquatic vegetation can negatively impact growth by reducing foraging success. Also, different fish prefer different types of habitats and will move to a new area if foraging conditions in their preferred location decline due to excessive growth of aquatic weeds.

An "optimal", one-size-fits-all fish habitat is impossible to describe, which leads to confusion and often erroneous conclusions. For example, a crappie fisherman has a different idea of a perfect habitat than does a bass fisherman. The parameters of an ideal habitat change based on the size and species of fish, the type of lake, structures present in the lake and numerous other factors. However, the "optimal" habitat that provides a beneficial environment for most animal populations is one that contains a large diversity of native plants.

Photo and illustration credits:

Page 7: Energy cycle; Horne and Goldman 1994
Page 8: Bluegill sunfish; Eric Engbretson, US FWS
Page 9: Largemouth bass; Dean Jackson, professional fisherman
Page 10 upper: Black crappie; Lawrence Page, Florida Museum of Natural History
Page 10 lower: Common carp; Richard A Bejarano, Florida Museum of Natural History
Page 11 upper: Northern pike; Robin West, US FWS
Page 11 lower: Yellow perch; Duane Raver, US FWS
Page 13: Low vs. high heterogeneity; Eric Dibble, Mississippi State University

1.3 Impact of Invasive Aquatic Plants on Waterfowl

Ryan M. Wersal: Minnesota State University Mankato, Mankato MN; ryan.wersal@mnsu.edu
Kurt D. Getsinger: US Army ERDC, Vicksburg MS; Kurt.D.Getsinger@usace.army.mil

Introduction

Studies that evaluate the relationship between waterfowl and aquatic plants (native or nonnative) usually focus on the food habits and feeding ecology of waterfowl. Therefore, the purpose of this section is to describe the dynamics of waterfowl feeding in relation to aquatic plants. The habitats used by waterfowl for breeding, wintering and foraging are diverse and change based on the annual life cycle of waterfowl and seasonal conditions of the habitat. For example, waterfowl require large amounts of protein during migration, nesting and molting, and they fulfill this requirement by consuming aquatic invertebrates. As noted in Sections 1.1 and 1.2, a strong relationship exists between high numbers of aquatic invertebrates and diverse aquatic plant communities, so diverse plant communities also play an important role in waterfowl health by hosting the invertebrates needed to subsidize waterfowl migration, nesting and molting. After all, waterfowl native to the US have evolved alongside diverse plant communities that are likewise native to the US and utilize these plants and associated invertebrates to meet their energy needs. Metabolic energy demands of waterfowl are high during the winter months, so waterfowl need foods that are high in carbohydrates such as plant seeds, tubers and rhizomes during winter. Many waterfowl will sometimes abandon aquatic plant foraging while on their wintering grounds and feed instead on high-energy agricultural crops such as wheat, corn, rice and soybeans.

The nutritional requirements of waterfowl have historically been met in shallow lakes and wetlands where diverse aquatic plant growth is abundant. It is therefore important to understand the interactions between waterfowl and aquatic plants in order to provide quality habitat throughout migration corridors. The abundance and availability of quality habitat with adequate food cover and water is the most important ecological component affecting waterfowl populations. In order to support waterfowl health, breeding and survival, the maintenance of quality habitats is crucial so that waterfowl have access to foods they prefer instead of having to feed on what is available.

The preferred food habitats and feeding ecology of waterfowl differ based on the group of waterfowl (i.e., dabbling ducks, diving ducks, or geese and swans). For example, dabbling ducks (also called puddle ducks) vary greatly in size and “tip up” during feeding. Their feeding is constrained by how far their necks can reach into the water column (12 to 18 inches) and depth of the water, so dabbling ducks prefer habitats with shallow water and/or moist soil. Diving ducks typically dive (as their name implies) to feed on benthic organisms such as clams and snails or to forage in sediments for tubers and rhizomes of aquatic plants. Geese and swans are the largest of the waterfowl and typically consume more plant material than dabbling ducks and divers; however, as the availability of natural habitats is diminished, geese and swans have shifted from primarily feeding in wetlands to extensive grazing in agricultural areas.

Dabbling (puddle) ducks

Dabbling waterfowl include such species as the mallard (*Anas platyrhynchos*), blue-winged teal (*Anas discors*), green-winged teal (*Anas crecca*), wood duck (*Aix sponsa*), gadwall (*Anas strepera*), northern pintail (*Anas acuta*), northern shoveler (*Anas clypeata*) and American widgeon (*Anas americana*). Most dabbling species are non-selective in their feeding habits and feed primarily on aquatic or moist-soil vegetation that is abundant in a particular location. Dabblers

alter their diets as necessary to take advantage of food resources that are available. Food selection by dabbling ducks often changes based on the season in order to maintain energy requirements of the bird. Protein is important during spring and summer for feather production and to ensure breeding success, so invertebrates are critical components in the diet of dabbling waterfowl during these seasons. In late fall and winter, dabblers consume plant material that is high in carbohydrates so they can maintain energy levels and generate body heat throughout the winter months. Dabbling waterfowl utilize submersed plant species as carbohydrate sources to fulfill their energetic demand. Most consume seeds as their primary food source, but some species (mainly widgeon and gadwall) use vegetative parts of plants as well. Also, the specialized bill structure of the shoveler, or spoonbill, allows for sifting and consumption of planktonic algae, which are also high in carbohydrates.

Submersed plant communities play important roles in the annual life cycle of dabbling waterfowl since these communities are a direct source of food and also serve as an environment that supports a diversity of aquatic invertebrates. The primary submersed aquatic plants consumed by dabblers are the native pondweeds (*Potamogeton* spp. and *Stuckenia* spp.), which produce fruits, seeds, starchy rhizomes and winter buds that are favored carbohydrate sources for dabbling waterfowl. Sago pondweed (*Stuckenia pectinata*) is reportedly one of the food plants most sought after by waterfowl. Sago pondweed is likely the single most important waterfowl food plant in the US and often accounts for a significant proportion of the food consumed by fall staging waterfowl, pre-molting waterfowl, flightless molting waterfowl and ducklings.

Diverse plant communities with a wide variety of submersed, floating and emergent plants have more architectural structure and habitat for invertebrates, which results in a greater selection of food sources for dabbling waterfowl. Water bodies that are infested with nonnative species such as hydrilla (Section 2.2), Eurasian watermilfoil (Section 2.3) and curlyleaf pondweed (Section 2.4) lack the habitat complexity required to support diverse invertebrate communities and are not preferred feeding areas for dabbling waterfowl. These nonnative species form dense canopies at the surface of the water, reduce native plant diversity and reduce the carrying capacity of the ecosystem. Also, if large portions of the littoral zones of several water bodies within an area are infested with nonnative plants, waterfowl may be required to continually move in search of adequate forage and resting areas. This constant movement results in poor body condition since high expenditures of energy impact wintering, migration and/or breeding fitness. Birds that are in poor body condition when returning to northern breeding grounds may have reduced nesting success or may not nest at all. If shallow wetlands and moist-soil areas become infested with invasive emergent weeds such as purple loosestrife (Section 2.16) or phragmites (Section 2.15), the quality of food and refuge habitat for ducklings and molting waterfowl could be diminished during summer months and could ultimately reduce survival. For example, ducklings and smaller species of dabbling waterfowl such as blue and green-winged teal feed in moist soil and in areas where water depths do not exceed 8 to 12 inches. As a result, dense infestations or monotypic stands of invasive weeds can limit foraging efficiency and food quality for these ducks.

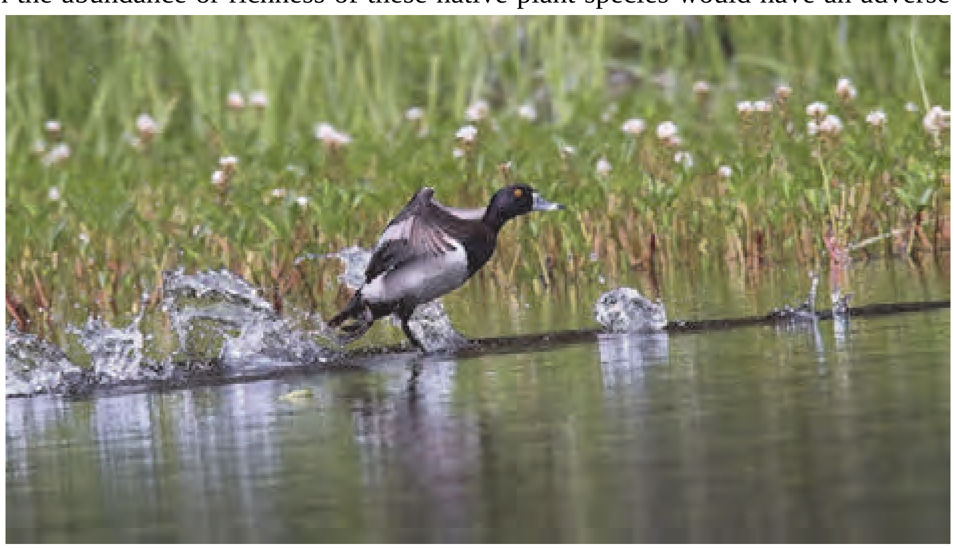
Diving ducks

Common diving ducks in North America include canvasback (*Aythya valisineria*), redhead (*Aythya americana*), lesser scaup (*Aythya affinis*), greater scaup (*Aythya marila*), ring-necked duck (*Aythya collaris*), bufflehead (*Bucephala albeola*) and common goldeneye (*Bucephala clangula*). Sea ducks and mergansers will not be discussed because sea

ducks spend much of their life cycle in marine environments and are rarely observed on inland waters and mergansers mainly consume fish.

The diet structure of diving ducks is similar to that of dabbling waterfowl because these ducks also rely on aquatic plants; their diet alternates with the annual life cycle of the birds and food selection is influenced by gender. Female diving ducks typically consume more invertebrates during nesting, incubation and brood rearing to maintain the protein and fat stores that result in good body condition. In contrast, male diving ducks (particularly older juveniles and adults) tend to consume more plant material. Canvasback ducks feed primarily on seeds and tubers of pondweeds and the native submersed plant vallisneria (*Vallisneria americana*), from which the bird takes part of its Latin name. Vallisneria is widely distributed and is considered the most important food source for canvasback ducks in northern states. Displacement of native vallisneria by invasive plants such as Eurasian watermilfoil or hydrilla will impact canvasback foraging behavior and can lead to annual fluctuations in canvasback populations. Canvasback numbers could decline or expand depending on the quality and abundance of vallisneria-dominated communities, which is linked to competition with invasive plants. Pondweeds are also very important food sources for redhead and ring-necked ducks, but these two species forage in shallow-water areas more frequently than other types of diving waterfowl and therefore consume a diversity of plant material. Ring-necked ducks feed heavily on wild rice (*Zizania palustris*), coontail (*Ceratophyllum demersum*), sedges (*Carex* spp.), rushes (*Scirpus* spp.), and the seeds and tender submersed shoots of the floating plant watershield (*Brasenia schreberi*). However, divers such as ring-necks are highly adaptive foragers and will reportedly feed on hydrilla tubers if hydrilla populations are abundant on their wintering grounds, particularly in large inland water bodies in Florida. The two species of scaup generally consume more invertebrates than plant matter, but plants do become important to scaup during fall and winter. With the exception of the ring-necked duck, all diving waterfowl will readily switch to feeding on mussels and clams in southern wintering grounds if plant material is limited.

Nonnative submersed weeds such as hydrilla, Eurasian watermilfoil and curlyleaf pondweed also have an impact on feeding activities of diving waterfowl. Since native pondweeds comprise a considerable portion of the food consumed by diving waterfowl, any reduction in the abundance or richness of these native plant species would have an adverse impact on waterfowl in that area. Diving waterfowl will reportedly consume the seeds of Eurasian watermilfoil and tubers of hydrilla; however, these observations were reported in areas heavily infested with these weeds and waterfowl were forced to forage on dense stands of these exotic plants, as their preferred native species were unavailable. It should also be noted that some propagules such as seeds can pass through the digestive tract of waterfowl and still be viable. Even if waterfowl utilize nonnative plants as food sources, this may



result in long-distance dispersal and spread of aquatic weeds to other areas of the country. Water bodies that are critical waterfowl habitat should be managed to promote the growth of a diverse native aquatic plant community because these are most utilized by diving waterfowl and they provide habitat for greater numbers and species of invertebrates.

Diving species of waterfowl also require emergent aquatic plants for nesting habitat. Canvasbacks and redheads nest almost exclusively above the water in specific types of vegetation. Hardstem bulrush (*Scirpus acutus*), cattails (*Typha* spp.), bur-reed (*Sparganium* spp.) and sedges that extend 1 to 3 feet above the water surface are preferred habitat for nesting. These plant species generally have more succulent and flexible stems that waterfowl can manipulate for nest construction. In contrast, nonnative plant species such as purple loosestrife and phragmites have hardened, woody stems that do not support waterfowl nesting. Purple loosestrife and phragmites will also outcompete native plants preferred for nesting, which further reduces breeding habitat that is becoming scarce due to pressure from human development and agricultural practices.

Geese and swans

Geese (Canada, snow and white-fronted) are primarily vegetarian and have shifted their feeding ecology toward agricultural grains and/or green-fields, including golf courses and parks. For example, corn and wheat have provided the majority of food for migrating and wintering Canada geese in recent decades and rice is frequently consumed by geese in the southern US. When agricultural grains become scarce in late winter, geese will feed on the green tissue of native moist-soil plants such as millets (*Echinochloa* spp.), smartweeds (*Polygonum* spp.), cut-grasses (*Leersia* spp.) and spikerushes (*Eleocharis* spp.). This switch in food sources also corresponds to times when crude protein is needed for migration and nesting. Swans are also primarily vegetarian but feed on aquatic plants more so than do geese. The diets of swans are based primarily on wigeongrass (*Ruppia maritima*), pondweeds and vallisneria during the winter months, but swans will forage in agricultural fields, golf courses or urban lawns when populations of aquatic plants are depleted.

Summary

Dabbling ducks, diving ducks, geese and swans are generalists and will consume the food sources available in a given area. Waterfowl prefer to forage and rest in shallow-water habitats that support diverse communities of submersed plants, including nonnative species. However, waterfowl usually prefer native species of aquatic and moist-soil plants to nonnative, invasive vegetation. Dabbling waterfowl prefer seeds of smartweed, millet, pondweeds, sedges and rushes, as well as invertebrates that typically thrive in association with these plants. Although waterfowl will utilize nonnative plants, these species are generally not preferred and are consumed only because they are locally abundant. Diving ducks such as canvasbacks and redheads rely heavily on pondweeds and vallisneria, but nonnative aquatic weeds such as Eurasian watermilfoil, hydrilla and curlyleaf pondweed can outcompete and reduce the presence of these valuable and desirable native plants. Furthermore, dense infestations of nonnative emergent species such as purple loosestrife and phragmites reduce the already-dwindling nesting habitat for many waterfowl species. North American waterfowl have evolved and thrive in habitats that support a variety of diverse native aquatic plants and management should focus on removing monotypic stands of nonnative plants to promote native plant growth.

Photo and illustration credits:

Page 15: Drake wood duck; Chuck Kartak Photography, used with permission
Page 16: Wintering mallard; Chuck Kartak Photography, used with permission
Page 17 upper: Incoming canvasbacks; Chuck Kartak Photography, used with permission
Page 17 lower: Flushing ring neck; Chuck Kartak Photography, used with permission
Page 18: Canada goose pair; Chuck Kartak Photography, used with permission

1.4 Impact of Invasive Aquatic Plants on Aquatic Birds

Mark V. Hoyer: University of Florida, Gainesville FL; mvhoyer@ufl.edu

Introduction

Birds that live at least part of their lives in or around water are referred to as aquatic birds and/or water birds. Each species has specific requirements that must be met in order to reproduce, survive, grow and reproduce again. It can be challenging to make broad statements that apply to all aquatic birds, but they are often grouped into subclasses based on habitat preference, which allows generalizations to be made about birds with similar requirements. Waterfowl are discussed in Section 1.3, but other groups of aquatic birds that use similar habitats include marsh birds, shorebirds and wading birds.

Marsh birds live in or around marshes (treeless wet tracks of grass, sedges, cattails and other herbaceous wetland plants) and swamps (wet, soft, low, water-saturated land that is dominated by trees and shrubs). This is a broad category that includes many unrelated species of birds, all of which prefer to nest and/or live in marshy, swampy areas. Marsh birds include herons, storks, ibises, flamingoes, cranes, limpkins and rails.

Shorebirds inhabit open areas of beaches, grasslands, wetlands and tundra. These birds, which include plovers, oystercatchers, avocets, stilts and sandpipers, are often dully colored and have long bills, legs and toes.

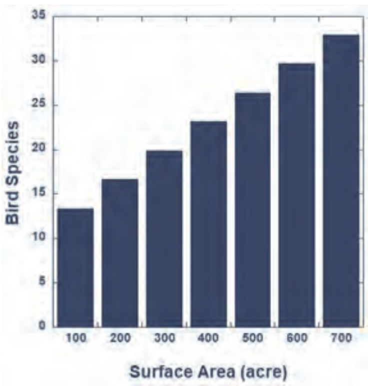
Wading birds generally do not swim or dive for prey, but instead wade in shallow water to forage for food that is not available on shore. Wading birds include herons, egrets, spoonbills, ibises, cranes, stilts, avocets, curlews and godwits. These birds generally have long legs, long bills and short tails, which allows them to strike and/or probe under the water for fish, frogs, aquatic insects, crustaceans and other aquatic fauna.

It is easy to see that some birds can fall into several of these general groups, so care should be taken when interpreting statements applied to birds in these groups. These subclasses group birds based on habitat preference, but birds are complex, adaptable animals. Thus, regardless of habitat, it may be possible to observe many different aquatic bird species if adequate food sources are available. The purpose of this section is to describe how aquatic bird populations are related to lake morphology, water chemistry and aquatic plants in lake systems and how the presence of large monocultures of exotic invasive plants such as hydrilla (Section 2.2) and phragmites (Section 2.15) may impact aquatic bird communities.

Lakes and aquatic bird communities

Birds are an integral part of all lake systems, but their role in the ecology of lakes has frequently been overlooked. This is surprising, since aquatic birds are often the first wildlife that is seen when visiting a lake and the vast majority of people who visit lakes enjoy their beauty and grace. However, most early research and management conducted on lake systems involved nutrient enrichment problems and aquatic plant management. The focus of this early research was primarily to provide potable water, flood control, navigation, recreational boating, swimming and fishing, and consideration was seldom given to aquatic bird communities that utilized these lakes. As a result, little information is available regarding how these different lake management activities affect aquatic bird communities.

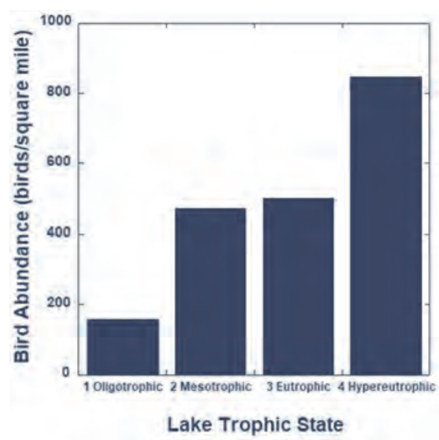
This situation began to change rapidly in the 1980s when many ornithologists (scientists studying birds) and limnologists (scientists studying freshwater systems) became increasingly conscious of the importance of birds to aquatic systems. These researchers have worked together to identify many significant relationships between lake limnology and aquatic bird populations, which can be used to predict the impact of habitat changes resulting from invasion by aquatic weeds and from lake management programs.



Lake area and aquatic bird species richness

There is a strong relationship between bird species richness (the number of bird species in an aquatic community) and the surface area of the lake they inhabit. Many studies have shown that plant and animal species richness increases as habitat area increases. Most researchers and lake managers agree that larger areas are more likely to include diverse habitats that allow more species niches. Based on this theory, the invasion of a lake system by an exotic species and the resulting monoculture of a single aquatic plant would decrease other environmental niches and would decrease the number of species of aquatic plants and ultimately aquatic birds using that lake system. However, there are few studies that document this type of impact aquatic weeds may have on bird populations.

Lake trophic state and aquatic bird abundance



Lake trophic state is the degree of biological productivity of a water body. Biological productivity generally describes the amount of algae, aquatic plants, fish and wildlife a water body can produce. The level of trophic state is usually set by the background nutrient concentrations of the geology in which the lake lies, because nutrients (primarily phosphorus and nitrogen) are the most common factors limiting growth of algae and plants that form the base of the biological food chain (Section 1.1). It is therefore not surprising that lakes with higher trophic states generally support more aquatic birds, since these lakes usually have an abundance of plants and animals that can be used for food and shelter by aquatic birds. Some question whether aquatic birds show up because a lake is productive or whether the lake becomes productive because birds bring nutrients into the system. There have been instances where large flocks of birds such as geese feed on terrestrial agricultural grains and then roost on a lake, ultimately causing elevated nutrient concentrations in a lake. However, most current research suggests that the majority of aquatic bird communities extract their nutrients from the lake and function more as nutrient recyclers than as nutrient contributors.

Most lake management efforts are directed toward the manipulation of lake trophic state, with most resources focused on reducing nutrients caused by anthropogenic activities. However, management agencies in some areas will actually add fertilizer (nutrients) in an attempt to increase productivity of plants, algae and fish, which increases angling activities. In either case, changes to the trophic state of a lake system have a corresponding impact on the aquatic birds that utilize the lake. If aquatic birds are an important component of an individual lake, this relationship needs to be considered before nutrient manipulations occur.

Aquatic plants and aquatic bird communities

Aquatic birds rely on aquatic plants to meet a large variety of needs during their life cycles. Some birds nest directly in aquatic plants, whereas others use plants as nesting material, foraging platforms, for resting and for refuge from predators. Aquatic plants are eaten by some bird species; in addition, some plants support attached invertebrates that are used as a food source by some aquatic birds. Since there are so many associations between the needs of aquatic birds and aquatic plants, it would be reasonable to expect a strong relationship between the abundance of all aquatic birds and the abundance of aquatic plants in a lake system. However, multiple studies have found no such relationship after accounting for differences in lake trophic state. This surprising lack of relationship between total bird abundance and total abundance of aquatic plants can be explained by the fact that individual bird species require different types and quantities of aquatic plants. Research has suggested that aquatic bird species can be divided into three general groups:

1) birds that are positively related to the abundance of aquatic plants
2) birds that are negatively affected by an abundance of aquatic plants
3) birds that have no relationship to the total abundance of aquatic plants but require the presence of a particular plant type for completion of their life cycle

However, these are loose generalizations and individual species of aquatic birds can transcend these plant groupings depending on a given lake system and the bird's life requirements.

Birds that are positively related to the abundance of aquatic plants. Many waterfowl, including the coots and ring-necked ducks described in Section 1.3, use aquatic plants as a food source and thus are generally more abundant in lakes with an abundance of aquatic plants. Other aquatic birds that prefer a habitat with plentiful aquatic plants include limpkins and curlews. These species are generalized feeders that consume insects, fish, small animals, snails and other aquatic fauna that are associated with aquatic vegetation. Limpkins and curlews are commonly observed walking on and foraging in floating aquatic plants, waterhyacinth (Section 2.11), salvinia (Section 2.13), native waterlilies (*Nymphaea* sp.) and other plants when this vegetation is present in densities sufficient to support the weight of the birds. If this type of habitat is not available, these birds will forage along sparsely vegetated shorelines and mudflats where water is shallow enough to allow wading. Birds in this group prefer lakes with an abundance of aquatic plants; however, these species will often locate and feed in more diverse habitats when their preferred environment is not available to them.

Birds that are negatively affected by an abundance of aquatic plants. Some birds such as snakebirds (*Anhinga anhinga*) and double-crested cormorants (*Phalacrocorax auritus*) must swim through the water to catch fish, crayfish, frogs and other aquatic fauna. Large amounts of aquatic vegetation interfere with the feeding ability of these aquatic birds; therefore, these birds tend to decrease in abundance when submersed weeds become abundant in a lake system. Other aquatic birds that prefer sparsely vegetated water are the threatened piping plover (*Charadrius melodus*) and the endangered interior least tern (*Sterna antillarum athalassos*). These species once fed, nested and were abundant on sandbars along the Missouri and Platte Rivers and in other similar areas in the central and northern US; however, piping plovers and interior least terns have experienced major population declines in the last 60 years. Dredging and damming of rivers has destroyed most of the sandbar habitat preferred by these species and flood control projects have reduced scouring and re-forming of new sandbars. In addition, old sandbars have become densely vegetated, further reducing the nesting and feeding grounds required by these aquatic birds. This is particularly problematic in the Midwest, where phragmites and purple loosestrife (Section 2.16) have invaded the majority of sandbars formerly inhabited by piping plovers and interior least terns.

Some aquatic birds are only affected by certain types of aquatic weeds. For example, eagles and ospreys soar over open water in search of fish swimming near the surface of the lake, so submersed aquatic weeds rarely hinder feeding by these species. In fact, since submersed plants reduce wind and wave action and improve water clarity, the presence of these aquatic plants may actually increase the feeding efficiency of sight feeders such as eagles and ospreys. However, dense populations of floating plants and floating-leaved plants (e.g., waterhyacinth, salvinia, waterlilies, etc.) may negatively impact the foraging success of sight-feeding aquatic birds because fish are hidden beneath the vegetation. Sight feeders may be forced to abandon lakes that are heavily vegetated with these types of plants and seek out new habitats with open water that provide an unobstructed view of their prey.

Birds that have no relationship to the total abundance of aquatic plants but require the presence of a particular plant type for completion of their life cycle. Some aquatic bird species – including the secretive American bittern (*Botaurus lentiginosus*) and least bittern (*Ixobrychus exilis*) – require tall, emergent vegetation like cattails (*Typha* sp.) and bulrush (*Scirpus* sp., *Schoenoplectus* sp.) for concealment from predators regardless of the total amount of aquatic vegetation present in the lake. Both species of bittern "freeze", with neck outstretched and bill pointed skyward, when danger threatens and sway in imitation of wind-blown emergent vegetation such as cattails. Even nestling least bitterns, still covered with down, adopt this posture when threatened. Invasion by exotic species of aquatic plants would probably not impact this type of bird species unless the exotic species reduces the abundance of the required emergent aquatic plants.

Many wading birds also fall into this group and do well in lakes regardless of the amount of aquatic plants, but one factor that may limit the success of these wading birds is the availability of water shallow enough for them to forage for food. Wading birds that inhabit lakes regardless of the abundance of aquatic plants include great blue heron (*Ardea herodias*), great egret (*Ardea alba*), snowy egret (*Egretta thula*), little blue heron (*Egretta caerulea*) and tricolored heron (*Egretta tricolor*). Larger wading birds can forage in water of greater depths, which increases the area available for foraging. Therefore, the great blue heron has an advantage over the smaller little blue heron in open water. However, larger wading birds may become tangled in vegetation when an invasive exotic species covers a lake; on the other hand, many of the smaller wading birds can actually wade on top of dense plant growth, which vastly increases their foraging area.

Summary

Aquatic birds come in an almost infinite number of sizes and shapes and require many different resources to complete their life cycles. A number of generalizations can be made regarding groups of similar bird types, but it is important to remember that all species are somewhat different. Also, individual species are adaptable and often able to use available resources even if those resources are not preferred. Encroaching invasive nonnative plants can increase, decrease or have little impact on a particular aquatic bird, which makes it difficult to predict the impact of aquatic plants on a given species. This dilemma becomes even more challenging when you consider that birds fly and can easily travel from lake to lake to find the habitat that best suits their needs, even though the distance may seem prohibitive.

Photo and illustration credits:

Page 19: Tricolor heron; Mark Hoyer, University of Florida
Page 20: Graphs; Mark Hoyer, University of Florida
Page 21: Red winged blackbird nest; Mark Hoyer, University of Florida
Page 22: Least bittern; Mark Hoyer, University of Florida

1.5 Aquatic Plants, Mosquitoes and Public Health

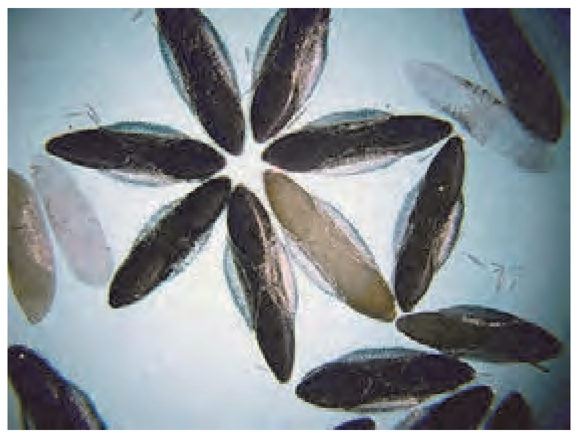
James P. Cuda: University of Florida, Gainesville FL; jcuda@ufl.edu

Introduction

Approximately 200 species of aquatic plants are classified as weeds in North America and nearly 50, or 25%, are considered to be of major importance. Aquatic plants become weedy or invasive when they exhibit rapid growth and produce dense monocultures that displace more desirable native plants, reduce biodiversity, interfere with flood control, impede recreation and navigation and create breeding sites for disease-vectoring mosquitoes.

Mosquitoes are insects that belong to the family Culicidae in the order Diptera, or true flies. They are similar in appearance to other flies except they have fragile bodies and their immature stages (eggs, larvae and pupae) develop entirely in aquatic environments. These insects are serious pests that have plagued civilizations throughout human history. In addition to their annoying and often painful bites, they transmit pathogens that cause some of the world's most devastating diseases, including chikungunya, dengue, West Nile, yellow and Zika fevers, encephalitis, malaria, and dog heartworm. Mosquito-transmitted viruses are commonly referred to as arthropod-borne or arboviruses. According to a recent report from the University of Florida, more than 500 million new cases of malaria are reported worldwide each year, resulting in about 1 million deaths. Most of the deaths that are caused by malaria are in children under 10 years of age. The importance of mosquitoes from a nuisance and public health perspective cannot be overstated.

Chikungunya



Chikungunya virus (CHIKV) is transmitted to humans by mosquitoes and the illness that occurs following successful transmission of CHIKV is called chikungunya. Chikungunya epidemics have been documented in India, Africa and Southeast Asia. Symptoms include fever, chills, headache, rash and severe joint pain with or without swelling, and the ankles and wrists are most commonly affected. Travel and globalization have increased the number of outbreaks in other regions wherever the mosquitoes capable of transmitting CHIKV are present. The yellow fever mosquito (*Aedes aegypti*) and Asian tiger mosquito (*Aedes albopictus*) are competent vectors that are able to carry and spread CHIKV. Both species are well-established in Florida, where they caused 11 cases of local transmission of chikungunya in 2014. Fortunately there were no further reports of local transmission by the end of 2016. In the event of an outbreak of chikungunya, mosquito control and public health agencies must identify source populations of the mosquito vectors in the affected areas and target them for control.

Dengue

Dengue is a viral disease and is often referred to as "breakbone fever". Symptoms of this mosquito-transmitted disease include headaches, high fever, rash, backache and excruciating joint pain, which gives rise to the common name. Disease symptoms usually occur about a week after a susceptible human has been bitten by an infected mosquito and dengue rarely results in death. However, because four strains of dengue virus are recognized, exposure of a previously infected individual to a different strain of dengue virus may result in a more severe case of dengue known as dengue hemorrhagic fever (DHF). There has been an increase in the incidence of DHF in the Western Hemisphere during the last 25 years, with outbreaks occurring in the Caribbean region, but ideal conditions for dengue transmission are also present in the southern US. The virus often is "imported" by people entering the country from the tropics. Also, the potential mosquito vectors (yellow fever and Asian tiger mosquitoes) are commonly found in close association with humans since they breed in natural and artificial water-holding containers near homes and businesses.

Yellow fever

Like dengue fever, the yellow fever virus is transmitted primarily in urban areas by the container-breeding yellow fever and Asian tiger mosquitoes. However, the effects of yellow fever on humans are more severe than those of dengue fever and during outbreaks, the human fatality rate often exceeds 50% of the affected population. Fortunately, the yellow fever virus is restricted to parts of Africa and South America. The likelihood of the yellow fever virus causing an epidemic in the US is extremely low for several reasons. First, yellow fever is a quarantinable disease; the Centers for Disease Control and Prevention in Atlanta continually monitor disease outbreaks in the Western Hemisphere. Second, travelers planning to visit parts of Africa and South America where the virus is endemic are vaccinated to prevent infection. Finally, humans moving to virus-free areas from locations where the virus occurs naturally are required to be vaccinated to prevent transmission.

Zika

Zika (ZIKV) is another mosquito-transmitted virus primarily in tropical regions where several epidemics have occurred during the last decade. Native to West Africa and Uganda, ZIKV typically cycles between infected forest tree hole mosquitoes and their primate hosts. ZIKV is unique in that local infections also can occur via sexual transmission. Symptoms of mild infections include rash, fever, joint pain and conjunctivitis and until recently, human infections were regarded as incidental and unimportant from a medical perspective. However, a strain of the virus traced to French Polynesia was discovered in northeastern Brazil in 2015 and the increased incidence of microcephaly in babies born to Zika-infected mothers was cause for concern. Local transmission by the yellow fever and Asian tiger mosquitoes was subsequently documented in most tropical countries of the Americas and in the United States, including Florida and Texas. Molecular diagnoses of Zika are recommended to confirm infection because of the similarity of Zika clinical symptoms to other mosquito-borne viral infections. Because no vaccine is available to prevent Zika infection, the Centers for Disease Control and Prevention advise pregnant women not to travel to locations where there is active transmission of the virus.

Encephalitis

Encephalitis means inflammation of the brain and is a disease of the central nervous system. Although there are several possible causes for encephalitis, one of the most important involves mosquitoes. There are seven major types of arboviral encephalitis in the US: California encephalitis (CE), Eastern equine encephalitis (EEE), Highlands J virus (HJ), St. Louis encephalitis (SLE), Venezuelan equine encephalitis (VEE), Western equine encephalitis (WEE) and West Nile virus (WNV). These viruses are normally diseases of birds or small mammals and each is caused by a different virus or virus complex. Humans and horses are considered “dead end” hosts for these viruses since there is little chance of subsequent disease transmission back to mosquitoes. However, human and horse cases of arboviral encephalitis range from mild to severe, with permanent damage to the central nervous system or even death. Mosquitoes involved in the transmission of arboviruses include species of *Aedes*, *Anopheles*, *Culex*, *Culiseta*, *Ochlerotatus*, *Coquillettidia* and *Psorophora*.

Malaria

Malaria was endemic in the US until around 1950 when window screens, air conditioning and mosquito control efforts essentially eliminated malaria in this country. Malaria is caused by four species of a protozoan parasite in the genus *Plasmodium*. This parasite, which is transmitted by a mosquito bite, destroys red blood cells and causes fever, chills, sweating and headaches in infected humans. If not treated, individuals that are infected with malaria may go into shock, experience kidney failure and eventually slip into a coma and die. The disease is transmitted by several species of *Anopheles* mosquitoes, which are permanent water mosquitoes (see page 25). These species are widespread and are most abundant from early spring (April) to early fall (September). Until recently, reported cases of malaria in the US were from travelers and returning military personnel who contracted the disease outside the country. However, cases of malaria occur periodically in the US when indigenous *Anopheles* mosquitoes transmit the disease from an infected human who traveled abroad to an uninfected human.

Heartworms

The filarial nematode (microscopic worm) *Dirofilaria immitis* is responsible for dog heartworm, a serious mosquito-transmitted disease that affects all breeds of dogs. Although the disease occurs in temperate regions of the US, it is more of a concern along the Atlantic and Gulf Coasts from Massachusetts to Texas. If left untreated, the infection rate in dogs can range from 80 to 100%. Foxes and coyotes probably serve as reservoirs for the disease. Cats and humans also can be infected but the parasite is unable to complete its development in humans. Mosquitoes in most of the common genera, including *Aedes*, *Anopheles*, *Culex*, *Ochlerotatus*, *Mansonia* and *Psorophora*, are capable of transmitting the disease. The life cycle of dog heartworm begins when an infected mosquito feeds on a dog. Juvenile worms (microfilariae) emerge from the mouthparts of the feeding mosquito and enter the dog's skin. The worms migrate in the muscle tissue for 3 to 4 months, penetrating blood vessels and eventually making their way to the right ventricle of the dog's heart, hence the name "dog heartworm". The worms reach maturity in around 5 months; adult female worms measure about 1 foot in length whereas males are only 6 inches long. The life cycle is completed when the adult female produces microfilariae that circulate in the blood and are ingested by a mosquito during a blood meal. Medication for preventing dog heartworm is available from veterinarians.

The role of aquatic plants in mosquito outbreaks

The aquatic stages of most mosquitoes are not adapted to life in moving waters. They require quiet pools and protected areas where they can obtain oxygen at the water surface or from plant roots via a single air tube (or siphon) in the larval stage or two tubes (or horns) in the pupal stage. Aquatic weed infestations create ideal habitats for mosquito development because the extensive mats produced by many weeds reduce the rippling effect of the water surface. Some mosquito species even have a modified air tube that they insert into the roots of aquatic plants to obtain oxygen. This protects them from light oils that are applied to the water surface for mosquito control.

From a mosquito control perspective, there are two major larval habitat categories that are of concern to aquatic plant managers: standing water (permanent and temporary) and flood water (detention and retention areas). Permanent water mosquitoes (e.g., species in the genera *Anopheles*, *Culex*, *Coquillettidia* and *Mansonia*) are associated with aquatic plants in freshwater marshes, lakes, ponds, springs and swamps. Temporary water mosquitoes (e.g., species in the genera *Culiseta*, *Ochlerotatus* [=*Aedes*] and *Psorophora*) are associated with vegetation in saline or brackish ditches, borrow pits and canals and freshwater drainage ditches, which alternate between wet and dry based on water use and rainfall events.

Permanent water

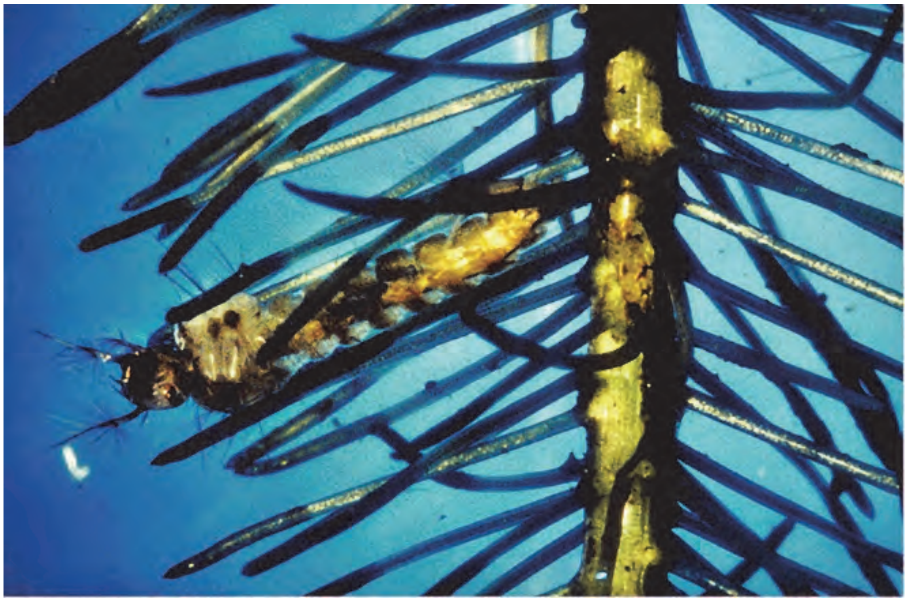
The amount and type of vegetation occurring in a permanent water body is a good indicator of its potential to produce mosquitoes. For example, the presence of floating mats of cattails (*Typha* sp.), torpedograss (*Panicum repens*), alligatorweed (*Alternanthera philoxeroides*) or paragrass (*Urochloa mutica*) suggest that larvae of permanent water mosquitoes are likely to be present. Also, dense stands of aquatic plants create ideal conditions for mosquito development by restricting water flow in drainage and irrigation ditches.

Flood water

Detention and retention systems are artificial ponds designed to capture floodwater from rainstorm events and filter it before it enters natural systems. Construction of stormwater detention/retention areas has increased dramatically throughout the US and they are often required by law for new commercial and residential developments. Detention ponds differ from retention ponds by the length of time they are “wet”. Detention ponds dry out only during drought conditions, whereas retention ponds are designed to dry out rapidly, usually within 72 hours. Under the right conditions, both types of flood control systems can produce aquatic vegetation capable of fostering mosquito outbreaks. Unless they are properly managed, detention/retention areas overgrown with aquatic vegetation can lead to serious mosquito problems. Detention ponds normally do not produce many mosquitoes unless they alternate between the wet and dry cycles that are required to produce floodwater mosquitoes. However, if they are not properly managed, they are often invaded by floating and rooted aquatic plants. The only way to prevent mosquito problems in residential and commercial detention/retention areas that contain these mosquito-producing plants is to control the plants.

Mosquitoes associated with specific aquatic plants

Some species of mosquitoes are associated with certain species of aquatic plants. For instance, the permanent water mosquito species *Coquillettidia pertubans*, *Mansonia dyari* and *M. titillans* are always associated with waterlettuce (Section 2.12), waterhyacinth (Section 2.11) and cattails (*Typha* spp.). The extensive fleshy root systems of these species provide an ideal substrate for larvae to attach and obtain oxygen through air tubes they insert into the plant roots. The roots of cattails and other plants also afford mosquito larvae some measure of protection from predators (including fish), as they are hidden from them. Other plants are good indicators of areas likely to produce floodwater mosquitoes. For example, sites with grasses, sedges and rushes often host enormous numbers of *Psorophora* mosquitoes that are vicious biters. On the other hand, the presence of extensive mats of duckweed (Section 2.14) or salvinia (Section 2.13) is indicative of low mosquito production areas. Although the root system of salvinia is highly branched, this floating aquatic fern is not a preferred host for mosquito larvae.



Summary

The association between aquatic plants and certain species of mosquitoes has evolved over millions of years. The uncontrolled growth of invasive plants often provides an undisturbed habitat that mosquitoes prefer and in which they can proliferate. Mosquitoes can colonize virtually any type of water body and aquatic vegetation provides a perfect environment for mosquitoes to thrive. Management of dense surface-growing nonnative and native aquatic plants in permanent and temporary water systems is critical to reduce the habitats suitable for mosquito development. After all, “…Without aquatic plants, most of our freshwater mosquito problems would not exist…” (Wilson 1981).

Photo and illustration credits:

Mosquito life stages photos (all from University of Florida IFAS Medical Entomology Laboratory)

- Page 23 upper: *Anopheles quadrimaculatus* eggs; Roxanne Connelly
- Page 23 lower: *Culex salinarius* larva; Michelle Cutwa-Francis
- Page 24: Mosquito pupa; James Newman
- Page 25: *Culex quinquefasciatus* adult; James Newman

Page 26: Mosquito larva attached to root of waterlettuce; T. Loyless, Florida DACS

2.1 Introduction to the Plant Monographs

William T. Haller: University of Florida, Gainesville FL (emeritus); whaller@ufl.edu

A small percentage of the thousands of plants introduced to the United States in the past three centuries have become problematic. Corn, soybeans, peanuts, potatoes, citrus, rice and many other introduced species are invaluable to human society. Regardless, all of the weeds described in this Section have one thing in common: they were introduced from other geographical areas by humans. Some native plants such as phragmites and bananalily have hybridized with introduced varieties or species to become more aggressive (see Sections 2.15 and 2.9 for more information). These weeds have caused significant economic and ecological damage to our aquatic resources. Millions of dollars are spent annually to reduce these impacts and similar efforts will continue into the foreseeable future. Lake homeowners associations and other concerned citizens should learn to recognize these invasive weeds and make every effort to prevent their introduction.

The authors of the plant monographs have devoted years to studying the biology and management of these plants. Each plant has distinct ecological and physiological characteristics that causes it to be invasive and some are more aggressive than others. A better understanding of how and why they are invasive may allow scientists to make better recommendations for how to prevent other invasive plants from being introduced into the US. If this manual was being written in 1950 it would likely include only five or six species: Eurasian watermilfoil (Section 2.3), curlyleaf pondweed (Section 2.4), waterchestnut (Section 2.10), waterhyacinth (Section 2.11), waterlettuce (Section 2.12) and possibly egeria (Section 2.5). At that time, these were regional problems (some still are) and did not occur over large geographical areas like hydrilla (Section 2.2), watermilfoil, phragmites and purple loosestrife (Section 2.16) do today. Starry stonewort (Section 2.7), monoecious hydrilla and floatingheart are relatively new and are likely to become more widespread in the future, while other plants such as waterlettuce, waterhyacinth, curlyleaf pondweed and phragmites have likely reached their geographical limits pending future climate change.

There are an infinite number of websites and other sources of information on invasive weeds available to the public. State natural resource agencies, county extension offices, local NGOs such as The Nature Conservancy, Sierra and Audubon Clubs and others usually have local members or staff with knowledge of invasive plants in your area. There are aquatic weed management companies throughout the US with expertise in plant identification and lake or pond biology.

The following websites are good sources of information; additional resources for each Section of this handbook are listed in the "For more information" Section beginning on page 205.

National scope

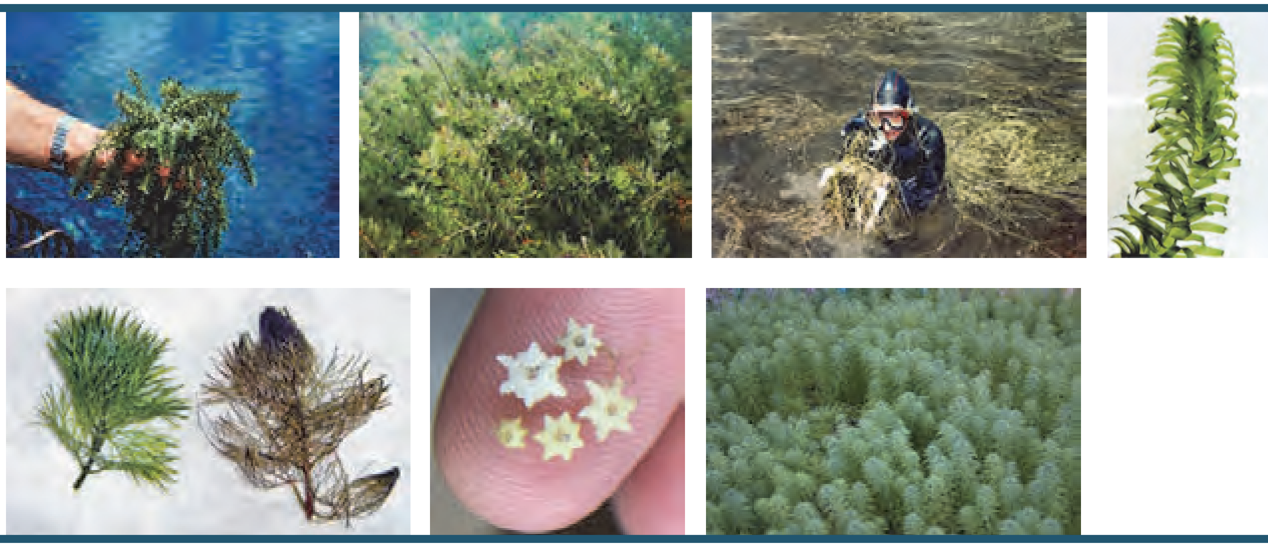
North America Invasive Species Management Association: www.naisma.org
University of Georgia: www.bugwood.org
US Army Corps of Engineers: https://apcrp.el.erdc.dren.mil
US Department of Agriculture The Plants Database: http://www.plants.usda.gov
US Environmental Protection Agency: http://www.epa.gov
US Geological Survey: https://nas.er.usgs/taxgroups/plants

Regional scope

University of Florida Center for Aquatic and Invasive Plants: http://plants.ifas.ufl.edu

North Carolina State University: https://aquaticweeds.wordpress.ncsu.edu
Texas A & M University: https://aquaticplant.tamu.edu
University of Minnesota: https://www.maisrc.umn.edu

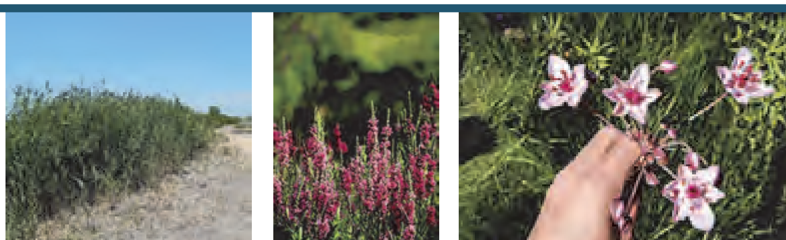
Submersed



Floating-leaved

Floating

Emergent



Algae

2.2 Hydrilla

William T. Haller: University of Florida, Gainesville FL (emeritus); whaller@ufl.edu
Rob Richardson: North Carolina State University, Raleigh NC; rob_richardson@ncsu.edu

Hydrilla verticillata (L.f.) Royle; submersed plant in the Hydrocharitaceae (frog's-bit) family
Derived from *hydr* (Greek: water) and *verticillus* (Latin: whorl) “water plant with whorls of leaves”

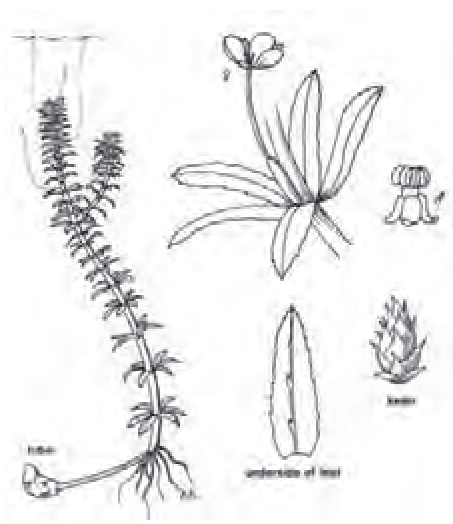
Dioecious introduced from Asia to Florida in the late 1950s
Monoecious introduced in 1970s in Mid-Atlantic states
Present throughout the southeast and north to New England and Wisconsin; west to California, Washington and Idaho

Introduction and spread

Hydrilla (*Hydrilla verticillata*) is the only recognized species in the genus *Hydrilla* but numerous biotypes occur in its Asian native range. Some biotypes are monoecious with separate pollen-bearing (“male”) and seed-producing (“female”) flowers on the same plant, whereas others are dioecious (each plant bears only “male” or “female” flowers). The monoecious and dioecious biotypes are almost identical and can be virtually impossible to tell apart. It appears that hydrilla was introduced to North America on at least two separate occasions which accounts for the two different biotypes present in the US. Hydrilla presently occurs in North America from Texas north to Wisconsin and eastward to Maine and in the west from Arizona north to Idaho. It was likely introduced through the aquaculture and aquarium trade. Hydrilla was added to the Federal Noxious Weed List in the late 1970s and is on many state prohibited lists as well. These listings prohibit interstate sale and shipment of the species, but hydrilla is readily available for purchase on the internet. It is easily spread by irresponsible boaters and others who move plants from one watershed to another, since the species reproduces and forms new colonies from small plant fragments that hitchhike on boats and other equipment. There is no direct evidence to suggest that hydrilla is spread by waterfowl or other aquatic fauna, but this type of transfer may occur between bodies of water that are in close proximity to one another. However, many confirmed initial infestations have occurred near public access points, suggesting that boaters continue to inadvertently transfer hydrilla on trailered boats.

Description of the species

Hydrilla is a rooted, submersed perennial monocot that grows in all types of bodies of freshwater, with growth limited only by water depth and velocity of flow. The stems of hydrilla are slender (about 1/32 of an inch in thickness), multi-branched and up to 25 feet in length – apical meristems can grow as much as an inch per day and there are dozens produced as the plant nears the water surface. Hydrilla forms dense underwater stands and often “tops out” to form dense canopies or mats on the surface of the water. All vegetative parts of hydrilla are submersed and the appearance of the species can vary drastically depending on growth conditions such as water pH, hardness and clarity.



Hydrilla has small (to 5/8 of an inch in length), strap-like, pointed leaves. The midrib on the underside of the leaf often has one or more sharp teeth along its length and leaf margins are distinctly saw-toothed, especially in hard water. Leaves are attached directly to the stem and are borne in whorls of four to eight around the stem, with a space of 1/8 to 2 inches between whorls. Healthy leaves are bright green, whereas leaves under stress from fungi, bacteria and sun-bleaching may be brown or yellow. Hydrilla is often confused with native elodea (*Elodea canadensis*) and exotic *Egeria densa* (commonly called egeria or Brazilian elodea) (Section 2.5). While these three species are very similar in appearance, leaves of native elodea are borne in whorls of three and those of egeria are arranged in whorls of four or five. In addition, only hydrilla has saw-toothed leaf margins; the leaf margins of the other species are smooth. It is often difficult – even for trained biologists – to tell hydrilla, native elodea and egeria apart. This makes early detection and rapid response efforts very difficult since by the time hydrilla is positively identified it has often produced reproductive tubers and turions. Plants can be positively identified as hydrilla by digging 3 to 4 inches into the soil and looking for the presence of tubers or turions among the roots, as hydrilla is the only one of these species to produce these reproductive structures.

Reproduction

Hydrilla is spread primarily by vegetative means (plant fragments) since each leaf node has axillary buds in the leaf axils capable of producing a new plant. Its spread by this method has been rapid and has increased the species' range throughout most of the southeastern US. Hydrilla produces two types of vegetative reproductive structures: turions and tubers. Turions are small (to 1/4 inch in diameter), cylindrical, dark green and borne in leaf axils, whereas tubers are larger (to 1/2 inch in diameter), potato-like, yellowish and attached to the tips of underground rhizomes 2 to 4 inches below the surface of the sediment. Dioecious hydrilla produces tubers and turions during winter short-day conditions in the southeastern US, whereas monoecious hydrilla behaves like a herbaceous perennial and produces these structures in mid to late summer in northern waters. Hydrilla is the only species in the Hydrocharitaceae family to produce tubers and turions, so the presence of these structures is considered confirmation that the plant in question is indeed hydrilla. Underground tubers can remain dormant for many years; this protects the species from management efforts such as drawdowns (Section 3.4) and allows plants to survive adverse conditions. Studies have shown that a single sprouting tuber of hydrilla planted in shallow water can produce several hundred tubers per square foot each year. Monoecious hydrilla is able to produce seeds, but their contribution to population development and expansion in thought to be negligible.

Problems associated with hydrilla

Hydrilla grows almost entirely underwater as a submersed aquatic plant and its growth potential is limited primarily by water clarity and depth of light penetration. Hydrilla has been reported at depths of 35 to 40 feet in crystal clear spring water and is commonly found at water depths of 15 to 20 feet in lakes with clear water. Hydrilla is uniquely adapted to grow under low light conditions, which allows it to colonize water that is deeper than most native submersed species can tolerate. For example, native submersed plants typically colonize the margins of shallow lakes where water depth is 6 to 8 feet. Hydrilla competes with native plants in these shallow areas, but also grows in much deeper water with little or no competition, which greatly extends the area of the vegetated littoral zone outward from the shoreline into deeper waters.

Hydrilla infestations often go unnoticed until the species tops out and reaches the surface of the water, where it forms hundreds of lateral branches due to the increased light intensity. This surface canopy or mat formed in the upper 1 to 2 feet of water comprises as much as 80% of the biomass of the plant on an area basis and limits light availability to lower-growing native submersed plants, which reduces species diversity over time. The ecological effects of this dense growth on the water surface include significant changes in water temperature, wave action, oxygen production, pH and other parameters, which reduce the suitability of infested waterways for use by aquatic fauna. Human activities are adversely affected as well – recreational use of water is limited, property values are diminished and there are increased public health and safety concerns (e.g., mosquito control, drowning, flooding). The severity of problems caused by hydrilla depends on the characteristics of the infested water body. An acre or two of hydrilla in a 100-acre lake may cause few problems; however, coves, bays or lakes with infestations of 80% or greater are significantly impacted by hydrilla.

Management options

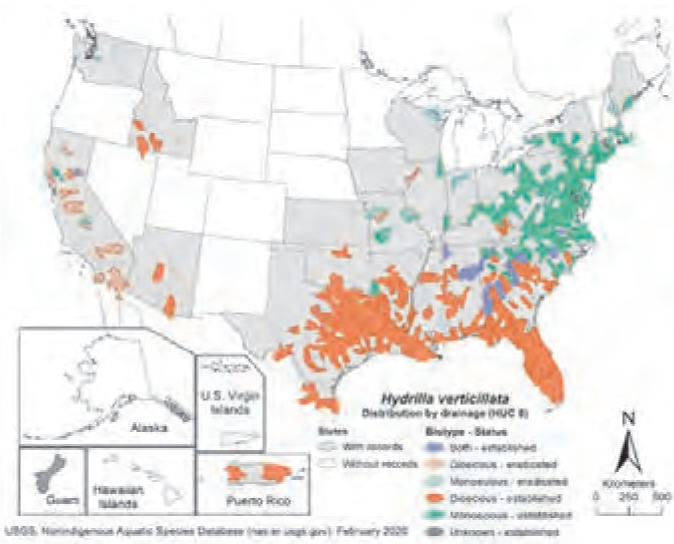
Clearly, preventing hydrilla from entering a water body is the best method to control this noxious species. Federal and state authorities have made it illegal to sell and transport hydrilla, which reduced this source of infestation. However, hydrilla still manages to increase its range and to colonize new bodies of water. Once hydrilla becomes established in a water body, control options are costly and generally must be employed on an annual basis.

Mechanical (Section 3.5) or physical (Section 3.4) control projects such as hand removal, benthic barriers or mechanical harvesters should be designed to prevent the spread of hydrilla fragments to other parts of the water body. Of course, if a lake is already extensively infested by hydrilla, there is less concern regarding plant fragmentation. Hand removal is labor-intensive and must take into consideration the presence of tubers and turions in and on the sediment, since failure to remove these structures virtually assures rapid reinfestation of the site. Mechanical harvesting can be expensive and most harvesters only cut to a water depth of 5 feet (although new deep-water harvesters are now available – see page 135). Since hydrilla meristems can grow an inch per day, control may only last for 2 to 3 months after mechanical harvesting. Another problem associated with mechanical harvesting is disposal of the harvested hydrilla. This vegetation has been evaluated for its potential as mulch, cattle feed, biofuel production and other uses, but its utility is very limited. Also, submersed plants do not produce much dry matter – a surface mat of hydrilla may weigh as much as 15 tons per acre, but contains only 5% (1,500 pounds) dry matter. As a result, harvested hydrilla is generally disposed of in a landfill due to its high water content (95% by weight) and low production of biomass. Drawdowns and freezing of hydrilla tubers and turions may provide temporary control in northern locations, but these measures provide only a season or partial season of control in the southeastern US. Thus, most hydrilla management programs rely on the use of biological control agents (grass carp) or herbicides.

Classical insect-based biocontrol of hydrilla has been studied for at least 50 years (Section 3.6.1). Researchers continue to seek possible biocontrol insects, pathogens and other agents in Asia and Africa. A few promising candidate insects have been discovered, studied and released to control hydrilla, but these insects have provided only localized and temporary reductions in hydrilla populations and are not considered to be viable biocontrol agents. In contrast, sterile triploid grass carp (Section 3.6.2) are widely used for hydrilla control in some states. Grass carp are released primarily in closed ponds or lakes and are sometimes used in conjunction with herbicides. Grass carp are not species-specific as required for the introduction of biocontrol insects; grass carp may prefer hydrilla but will consume most submersed and emergent aquatic plants. As a result, most states regulate the stocking and use of grass carp. Despite this challenge, grass carp continue to be the most effective method for biological control of hydrilla where their use is legal and practical.

Several herbicides (Section 3.7.1) can be used to effectively control hydrilla, but one of the most significant problems associated with chemical control of any submersed species is dilution. An acre of water that is one foot deep comprises 325,800 gallons of water, which results in tremendous dilution of herbicides. In addition, water flow or movement greatly reduces the amount of time hydrilla is exposed to the herbicide. These factors can make it difficult to control hydrilla using chemical methods, so treatments have to be designed to take dilution and water movement into consideration to achieve optimum results. See Section 3.7.1 for concentration/exposure requirements for aquatic herbicides.

Fast-acting contact herbicides – including copper, diquat, endothall, florpyrauxifen-benzyl and flumioxazin – are taken up quickly by hydrilla and usually cause plant death and decay in a few weeks. Most contact herbicides are applied at concentrations of 200 or greater ppb, but the fast-acting contact herbicide florpyrauxifen-benzyl is used at very low concentrations (2 to 48 ppb). Contact herbicides are generally used for spot treatments, strip treatments along shorelines and in areas where water movement would limit the use of slower-acting systemic herbicides that require much longer exposure times. Slow-acting systemic herbicides – including fluridone, imazamox, penoxsulam, bispyribac and topramezone – control hydrilla by inhibiting enzyme activity. These herbicides are usually applied as whole-lake treatments and provide control of hydrilla only when a long period of contact or exposure is possible. An advantage to systemic herbicides is that they are effective at low doses (usually concentrations of less than 25 to 50 ppb of fluridone, penoxsulam, bispyribac and topramezone). These herbicides slowly kill plants by starving them over a long period of time, but usually provide one to two years of control. Slow plant decay resulting from systemic herbicide treatments minimizes possible oxygen depletion, which reduces the potential for fish mortality. The disadvantage of systemic herbicides is that they generally require a whole-lake treatment, or at least treatment in coves, bays and other areas where water movement and dilution are reduced and there is little or no water exchange. Most states require permits to apply herbicides in public (and some private) waters, so contact your state water authority for further advice and information. An additional valuable source of information is the herbicide label, which is available on the manufacturer website or at www.cdms.net and describes the safety requirements, how the herbicide works, how much to apply and other useful information.



Eradication efforts

Conservation agencies and resource managers are very concerned about the rapid spread of hydrilla and eradication efforts are underway in several states. Hydrilla has been successfully eradicated in primarily small ponds in Wisconsin, Washington, Missouri, Iowa and Indiana. California has had an eradication program in place for nearly four decades and has spent millions of dollars stocking grass carp, dredging, draining and using herbicides in their aggressive control efforts. Other states with ongoing eradication programs include Maine, Massachusetts, Connecticut, New York, Idaho and possibly others. These expensive and time-consuming efforts are undertaken to contain and prevent hydrilla from spreading to other state waters, where infestations would cause both economic and recreational losses, possible flooding and negative ecological impacts that have occurred in other states.

Dioecious hydrilla

The USGS map of watersheds that host populations of the two hydrilla biotypes clearly shows that dioecious "female" plants spread from their initial 1950s introduction to Florida northeast to Kentucky, North Carolina and west to California. The Iowa population (farm pond) was likely introduced from Florida and was eradicated in the 1970s. Idaho provides a classic example of how humans move plants over large geographical distances. Blessed with many geothermal springs, Idaho is a primary producer of warm-water fish such as tilapia and tropical ornamental species for the aquaculture market in the US. Dioecious hydrilla was found in several commercial warm-water aquaculture facilities in the Snake River basin, where it was possibly introduced via transport of live fish from Florida. It was also found in the warm springs of the Bruneau River along with waterlettuce (Section 2.12) plants that were obviously introduced in the spring by an unknown person who admired their ornamental qualities. In addition, dioecious hydrilla was discovered in a small spring-fed urban canal in Boise, where it was likely introduced via a dumped aquarium. All of these sites are under eradication orders by the Idaho Department of Agriculture with cooperation from landowners and no hydrilla has been found in the Snake River to date. The Idaho situation is unique and the question remains: will dioecious hydrilla grow in the cold waters of the northeastern states or upper Midwest? Time will tell!

Dioecious hydrilla in the Southeast spreads only via fragmentation or other vegetative means because only the dioecious "female" biotype has been found in the southern US; no pollen in available, so it is unable to produce seeds. This biotype produces underground tubers under short-day conditions from late September through April, but only when water temperatures are greater than 40 °F. Similarly, turions are formed primarily under short-day conditions. These structures are likely an adaptation to allow survival under adverse conditions such as alternating wet and dry periods in a monsoon climate. Turion production is greatly increased if the plants are floating fragments, but tubers are only formed on underground rhizomes of rooted plants. Dioecious hydrilla behaves like a perennial and grows throughout the entire year in the warm waters of southern Florida, Louisiana and Texas. In more northern areas (or following very cold winters), growth is greatly reduced; untreated mats of hydrilla sink to the bottom of the water body, where they remain until warm temperatures return and trigger new growth from the axillary buds of the previous year's sunken vegetation. The role of tubers and turions in initiating or contributing to early spring growth under these conditions is not well-understood, but these structures probably do not have a significant effect on early growth. However, tubers and turions are very important to the re-establishment and growth of hydrilla following successful herbicide treatments, drawdowns or other control techniques that destroy the stems of hydrilla. The longevity or dormancy of turions is thought to be relatively short (2 to 3 years), whereas tubers remain viable for considerably longer periods of time (perhaps 8 to 10 years or more). Resource managers have reported that some Florida ponds stocked with grass carp for hydrilla control 10 or 15 years ago have experienced a resurgence in hydrilla populations after the carp were removed or died off. The question remains as to whether the regrowth of hydrilla following many hydrilla-free years is the result of long-term dormant tuber sprouting or from re-introduction of hydrilla from an outside source.

Monoecious hydrilla

Monoecious hydrilla was reported almost concurrently near Washington DC and Raleigh NC in 1980, although both sites were likely infested for several years before plants were properly identified. This biotype may now be found as far south as Georgia and Alabama and as far north as Maine. It extends as far west as Nebraska, but has also been introduced into California. In 2019, a different biotype of monoecious hydrilla was reported from the Connecticut River, but this biotype has not yet been fully described in the literature and will not be further discussed here. Genetic analyses of the first monoecious introductions have shown that they are closely related and linked to hydrilla found growing near Seoul, South Korea. It has been speculated that this monoecious type could be a hybrid between the US dioecious biotype and another dioecious biotype, but data to support or refute this hypothesis are lacking. Monoecious hydrilla is typically smaller and finer than dioecious hydrilla and generally resembles elodea in size (dioecious hydrilla is more similar to the larger, coarser *Egeria densa*). This difference can be helpful for distinguishing between the two biotypes in the field, but are not completely reliable as many factors can influence the appearance of these plants.

There are significant differences in the life histories of the original US monoecious hydrilla (which typically behaves as a herbaceous perennial) and dioecious hydrilla (in which shoots typically overwinter in the southern portions of its range). Monoecious hydrilla tubers and turions sprout in spring, with most sprouting occurring within a one-month period, although a small percentage of tuber sprouting may occur through the summer months. It has been reported that half of monoecious hydrilla turions sprout by mid-June and half of tubers sprout by mid-July. Initial growth is lateral along the lake bottom, which may be due to having minimal competition from other submersed plants at the start of the growing season, then after plants are established, shoots begin to grow toward the surface of the water. Growth increases as water temperature increases; exponential growth occurs during the warmest portion of the growing season and dense monoecious stands may produce topped-out growth in late summer into fall. Tuber formation is stimulated by long summer days and continues into fall. Axillary turions form late in the growing season as shoots begin to senesce. Floral initiation occurs during midsummer, but flowering and potential seed production seem to be very minor concerns compared to the large amounts of tubers and turions produced.

The main challenge in long-term management of monoecious hydrilla is the presence of tubers. Axillary turions are also produced, but these are generally smaller and shorter-lived and are likely meant to remain viable for a single winter and sprout the following spring. Tubers have more stored carbohydrates, which allows for longer dormancy periods, and remain viable for seven or more years. Seed production can occur but is not often seen in the field and is likely an irregular or rare event. Also, the ability of different biotypes to hybridize is not well-understood. Colder temperatures seem to increase tuber sprouting, thus larger sprouting percentages have been observed in New York than in North Carolina, Georgia or Alabama. From a management perspective, this means that tuber banks in southern climates will require more time for depletion than will tuber banks in northern regions.

One of the first locations of monoecious hydrilla establishment in the US was in the Potomac River. However, monoecious hydrilla rapidly spread through reservoirs and lakes in many states and was generally considered a problem of lakes and impoundments. In the last few years, monoecious hydrilla has been reported in numerous flowing water systems, including the Erie Canal, the Ohio, Croton, Eno, Cape Fear and Chowan Rivers and others. Although the Potomac infestation is generally considered non-problematic, many of the other infestations (for example, Erie Canal, Croton and Eno Rivers) have been significant enough to warrant management even though it is much more difficult to implement management practices in flowing water. A major concern is that monoecious hydrilla will outcompete and displace native organisms. In the Eno River, for instance, hydrilla is overtopping and outcompeting native riffleweed (*Podostemum ceratophyllum*), which provides critical habitat to the rare Panhandle Pebble Snail, and was the impetus for hydrilla management in this ecosystem.

Summary

Prior to 1950 there was no scientific information suggesting that hydrilla would cause such serious problems throughout the world. Hydrilla has become one of the worst submersed weeds globally as water resources have been developed and now causes problems in all tropical and subtropical continents with the exception of Africa, where it is believed that native herbivorous fish (cichlids) apparently keep its growth in check. Dioecious hydrilla has spread from Florida north to Virginia and Kentucky and northwest to California and Idaho in the span of only 50 years. The annual cost to control hydrilla in public waters in Florida alone totals approximately $15 million. Florida is particularly impacted by hydrilla due to its moderate climate and shallow, naturally nutrient-rich lakes, but research on the distribution of hydrilla in Asia predicts that hydrilla could colonize virtually any area in North America and could survive as far north as Hudson Bay.

Both biotypes of hydrilla are best managed through the use of herbicides. Managers usually treat monoecious hydrilla in spring or early summer when this biotype is in its lateral growth stage and requires somewhat lower application rates to achieve control. However, some applicators have noted that monoecious hydrilla requires higher doses of diquat and copper compared to the amounts required for control of dioecious hydrilla. Eurasian watermilfoil (Section 2.3) was historically the major submersed weed problem in the Tennessee River Valley (TVA) reservoirs, but dioecious hydrilla became widely established in the 1990s and has taken over much of the area previously covered by Eurasian watermilfoil. Also, TVA biologists have noted that monoecious hydrilla is now replacing much of the dioecious biotype in their systems.

The search for biocontrol insects and other agents continues, but some believe that monoecious hydrilla might be less susceptible to insect biocontrol agents that the dioecious biotype because monoecious populations are topped out for only a few weeks during the year. Sterile grass carp do not seem to have a preference for either biotype.

The discovery of monoecious hydrilla in the 1970s and its subsequent survival and spread from the Mid-Atlantic States throughout New England may well prove the predictions of survival into northern Canada to be true. The similarity in appearance of the hydrilla biotypes makes positive identification impossible without confirmation by genetic analysis. Also, the similarity of hydrilla to other submersed plants makes early detection efforts very difficult. Hydrilla has now become the most widespread and costly submersed non-native weed in North America. The monoecious biotype is currently spreading rapidly into new areas, while the dioecious biotype has likely already colonized most areas suitable for its growth in the southern US. Monoecious hydrilla has replaced much of the dioecious hydrilla in the TVA system, but only time will tell whether this biotype will fail to colonize, co-exist with dioecious hydrilla, or outcompete dioecious hydrilla in the southern states.

Photo and illustration credits:

Page 29: Hydrilla infestation; Vic Ramey, University of Florida
Page 30 upper: Line drawing; University of Florida Center for Aquatic and Invasive Plants
Page 30 lower: Hydrilla bouquet; William Haller, University of Florida
Page 32: US Geological Survey, Nonindigenous Species Database, Gainesville FL. https://nas.er.usgs.gov/taxgroup/plants

2.3 Eurasian Watermilfoil

John D. Madsen: USDA Agricultural Research Service, Davis CA; jmadsen@ucdavis.edu

Myriophyllum spicatum L.; submersed plant in the Haloragaceae (watermilfoil) family
Derived from *myrios* (Greek: numberless), *phyllon* (Greek: leaf) and *spica* (Greek: spike) "plant with many leaf divisions that bears flowers in a spike"

Introduced to several locations in the US from Europe in the 1940s
Present throughout the continental US and Alaska

Introduction and spread

Eurasian watermilfoil (*Myriophyllum spicatum*) is one of fourteen species of *Myriophyllum* present in the US. Most species of this genus in the US are native, but two (*M. aquaticum* and *M. spicatum*) are exotic species that have been introduced to North America. Of these two exotic species, Eurasian watermilfoil is much more widespread and more problematic. The species was first reported in the US in the 1940s and spread rapidly into the mid-Atlantic and

midwestern states in the 1960s and 1970s. Eurasian watermilfoil also became a serious problem in the hydropower and flood control reservoirs of the Tennessee River, where large-scale applications of herbicides were used in an attempt to eradicate the weed. Eurasian watermilfoil is still present in the TVA (Tennessee Valley Authority) system but has largely been displaced by hydrilla (Section 2.2). More recently (from the 1980s until 2009) the species has invaded lakes in Idaho, Minnesota and Maine and continues to expand its coverage throughout the northern US. Eurasian watermilfoil is now the most widespread submersed aquatic weed in the northern half of the US.

Eurasian watermilfoil has been introduced to the US multiple times and was likely first brought to North America in ship ballasts or as an ornamental plant for aquariums or water gardens. Accidental spread of Eurasian watermilfoil within the US is due primarily to transportation of contaminated boat trailers, boat parts and bait containers, but the species is also spread through the aquarium trade. Once Eurasian watermilfoil is introduced to an aquatic system, it spreads prolifically by stem fragments that are produced both naturally (when stem sections detach from the plant at abscission sites) and as a result of mechanical breakage (when plants come into contact with boat motors and intense wave action). Some researchers speculate that Eurasian watermilfoil may be spread by wildlife or waterfowl; however, no direct evidence exists to support this theory. Eurasian watermilfoil produces numerous viable seeds, but the seeds contribute little to the propagation and spread of the plant. Eurasian watermilfoil was too widespread to be listed as a Federal Noxious Weed when the list was first developed; however, the species is listed on numerous state noxious and prohibited plant lists.

Description of the species

Eurasian watermilfoil is rooted in the sediment and grows completely underwater as a submersed plant that forms a dense canopy on the water surface. The species is commonly found in water from 1 to 15 feet in depth but can occur at depths of up to 30 feet if the water is extremely clear. Eurasian watermilfoil is an evergreen perennial plant that produces persistent green shoots throughout the year and overwinters as root crowns. Leaves are pinnately compound (feather-like), with each leaf composed of 14 to 24 pairs of leaflets arranged in whorls (groups) of four at the nodes of the stem. Stems and plant tips may appear reddish, but color is not consistent and may vary based on a number of factors, including environmental conditions. Flowers form on short aerial stems that hold them above the water and have both pollen-bearing (“male”) and seed-producing (“female”) flowers. Flowers are wind-pollinated and produce up to four nutlets per flower. Eurasian watermilfoil is difficult to identify and is often confused with several native species of *Myriophyllum*, including northern watermilfoil (*M. sibiricum*) and whorled watermilfoil (*M. verticillatum*). Hybridization between Eurasian and northern watermilfoils can occur in the field and the seedlings produced from these cross-pollinations often have features that are intermediate to the parental plants.

Reproduction

Eurasian watermilfoil produces a significant number of viable seeds and plants can be propagated from seed in the laboratory or greenhouse. However, successful colonization of new plants from seed in nature has not been documented. As a result, sexual propagation is generally thought to play an insignificant role in the spread of Eurasian watermilfoil. The species reproduces predominantly by vegetative means through fragmentation, which occurs when stems are broken mechanically (from wave action or contact with boat motors) and when stem sections naturally abscise or detach from the plant. Stem sections that result from natural breakage have high concentrations of starch and are likely responsible for most of the spread of the species. Eurasian watermilfoil can also spread by forming new root crowns on runners, which are produced when stems arch down, come into contact with the sediment and form roots that create a new root crown. Root crowns can also spread through the formation of rhizomes under the sediment, although detailed studies of this process have not been conducted. Root crowns overwinter and produce new shoots every year. As a result, more stems are added to root crowns each year, which increases stem density in the water column.

Problems associated with Eurasian watermilfoil

Because Eurasian watermilfoil grows entirely underwater as a submersed aquatic plant, the range of water depths the species can inhabit is limited by light penetration and water clarity. A dense canopy often forms at the surface of the water, which interferes with recreational uses of water such as boating, fishing and swimming. Dense growth of Eurasian watermilfoil may also obstruct commercial navigation, exacerbate flooding or clog hydropower turbines. In addition, excessive growth of the species may alter aquatic ecosystems by decreasing native plant and animal diversity and abundance and by affecting the predator/prey relationships of fish among littoral plants. A healthy lake is damaged because heavy infestations of Eurasian watermilfoil lower dissolved oxygen under the canopy, increase daily pH shifts, reduce water movement and wave action, increase sedimentation rates and reduce turbidity.

Management options

Prevention is always the best option to avoid infestations of Eurasian watermilfoil. Posting signs at boat launches and requesting that lake users watch for Eurasian watermilfoil and remove all plant material from boats before launching can be a successful strategy. When prevention methods are unsuccessful, early detection and rapid response to new infestations have been shown to reduce management costs over the long term.

There are currently no biological control agents that effectively control Eurasian watermilfoil. For example, grass carp (Section 3.6.2) do not prefer to feed on this species. Numerous studies have been conducted to evaluate the utility of native insect herbivores as potential biocontrol agents of Eurasian watermilfoil, but none have proven to be predictable and effective to date. Also, if native insects were able to effectively control introduced populations of Eurasian watermilfoil, new introductions of the weed would not result in population development and expansion to weedy proportions. Historical accounts of the introduction and spread of Eurasian watermilfoil suggest this has not occurred. In addition, the use of native insects as biocontrol agents remains controversial (Section 3.6).

Several herbicides can be used to effectively manage Eurasian watermilfoil. Contact herbicides – including diquat and endothall – provide good control, whereas systemic herbicides such as 2,4–D, florpyrauxifen-benzyl, fluridone and triclopyr provide excellent control. Herbicides should be selected based on site size and conditions, water exchange characteristics, potential water use restrictions, federal, state and local regulations and economic considerations (Section 3.7.1).

Mechanical controls (Section 3.5) are also widely used to control small infestations of Eurasian watermilfoil. Mechanical harvesting and raking provide temporary but fair control in bodies of water that are small to moderate in size, whereas hand harvesting and suction harvesting provide longer term control than mechanical harvesting or raking. None of these mechanical methods alone results in long-term control of Eurasian watermilfoil; as such, these methods should be employed as part of an integrated weed control strategy.

Physical control techniques such as drawdowns, dredging and bottom barriers (Section 3.4) can reduce or prevent growth of Eurasian watermilfoil by altering the environment. Drawdowns require dewatering of the affected lake or pond and are particularly effective during the winter. Draining the water out of the system exposes the root crowns of Eurasian watermilfoil to the air and results in desiccation and death of the plants. Dredging is expensive but results in water depths too great for plants to grow. Dredging provides multi-season control but should only be used as part of a broader lake restoration effort. Bottom barriers are semi-impermeable sheets of synthetic material that are placed over the plant bed, which kills the plants underneath. Bottom barriers are expensive but can provide effective control of Eurasian watermilfoil in small areas.

Summary

Eurasian watermilfoil is an exotic aquatic weed that is widely distributed throughout North America. The species is most commonly associated with problems in temperate lakes, but invades tidal estuaries, saline prairie lakes, rivers and southern reservoirs as well. Although the economic impact of Eurasian watermilfoil is not as great as that of hydrilla or waterhyacinth (Section 2.11), its geographic and ecological distribution surpasses that of other North American aquatic weeds. In fact, problems associated with Eurasian watermilfoil are significant enough that states such as Idaho, Minnesota, Vermont and Washington have developed specific management programs to control invasions of Eurasian watermilfoil.

Photo and illustration credits:

Page 35: Eurasian watermilfoil infestation; Ryan Wersal, Minnesota State University Mankato
Page 36: Line drawing; University of Florida Center for Aquatic and Invasive Plants
Page 37: Eurasian watermilfoil; John Madsen, USDA, Davis CA
Page 38: Eurasian watermilfoil; John Madsen, USDA, Davis CA

2.4 Curlyleaf Pondweed

Thomas Woolf: Montana Fish Wildlife and Parks Aquatic Invasive Species Bureau, Helena MT;
Thomas.woolf@mt.gov

Potamogeton crispus L.; submersed aquatic plant in the Potamogetonaceae (pondweed) family
Derived from *potamos* (Greek: river), *geiton* (Greek: neighbor) and *crispus* (Latin: curly) “curly-leafed plant close to the river”

Introduced from Europe in the mid 1800s
Present in all lower 48 states; particularly problematic in northern states and Canada

Introduction and spread

Native to Europe, Asia, Africa and Australia, the first known collection of curlyleaf pondweed in North America occurred in Philadelphia in 1841. The plant spread to the Great Lakes region in the early 1900s and today is found in

all of the contiguous 48 states. The spread of curlyleaf pondweed throughout the US can be attributed to boat and fish hatchery activity. Curlyleaf pondweed is now thoroughly naturalized in the United States and Canada and is considered an exotic weedy species throughout its range.

Description of the species

Curlyleaf pondweed is a rooted submersed herbaceous perennial monocot that grows in lake and river systems and aggressively outcompetes native submersed vegetation. The species has wavy leaves with finely serrated or toothed margins and a “crisp” leaf texture. Leaves are typically green early in the season and can become red when they near

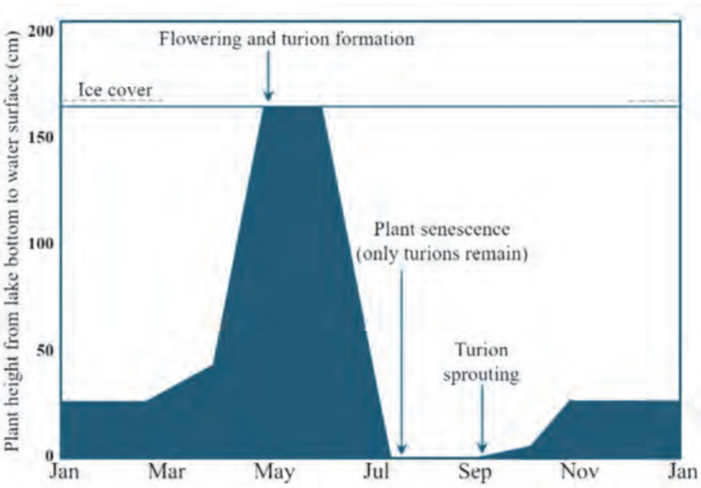
the water's surface. The oblong-shaped leaves are 1 to 3 inches in length and are attached to the stem in an alternate arrangement. Long spaghetti-like stems form as the plant quickly grows to the water's surface and develops into dense weedy mats.

Curlyleaf pondweed grows in conditions ranging from ice-covered waters with very low light intensities to summer conditions with very warm temperatures and intense sunlight. Colonization by curlyleaf pondweed is limited by light availability and the species typically inhabits waters that range from 3 to 6 feet in depth, but curlyleaf pondweed has been found at depths of more than 20 feet in very clear water. This species prefers to grow in still water, but curlyleaf pondweed is quite tolerant of flow and is found in many river systems throughout the US and Canada.

Curlyleaf pondweed is often found in nutrient-rich or eutrophic systems and the species has a high tolerance for nutrient pollution and low light conditions. In fact, the species is sometimes considered an indicator of pollution and eutrophication due to its tolerance of low light and high dissolved nutrients.

Reproduction

Curlyleaf pondweed reproduces primarily by producing turions and rhizomes. Turions are hardened modified reproductive buds that form from apical buds, in leaf axils or directly from rhizomes prior to plant senescence in early summer. A single plant produces an average of 5 turions, with each turion averaging 4 buds. Turions constitute over 40% of the total plant biomass prior to senescence and turion densities of more than 1,000 per square foot have been reported in lake sediments. Each turion can remain viable in the sediment for multiple seasons and can sprout multiple times. Flowering usually coincides with turion formation. Flowers are very small, inconspicuous and borne on small spikes that emerge above the water surface. Seeds are produced but germination rates are quite low (0.5%). As a result, reproduction of curlyleaf pondweed is due mainly to the production and sprouting of vegetative turions.



Curlyleaf pondweed has a life cycle that is fairly unique for submersed aquatic plants. Plants flower and produce turions, then die back or senesce typically in early summer. Turions lie dormant throughout the summer and then sprout in the fall when water temperatures drop to below 66 °F and daylength shortens to fewer than 11 hours of daylight. Plants grow and may reach from an inch to several feet in height until water temperatures fall below 50 °F. When temperatures drop below 50 °F, growth of curlyleaf pondweed slows or stops and plants overwinter in a very slow-growing or dormant state. Since the species overwinters with green growth above the sediment, curlyleaf pondweed often has an advantage over native species when growth resumes in the spring.

Curlyleaf pondweed can grow up to 4 inches per day when days become longer and water temperatures start to rise in early spring. Plants quickly grow to the surface and turion production and flowering begins. Dense mats of curlyleaf pondweed can form on the water surface and shade out competing species. Turion production and flowering are followed by senescence or dieback, which occurs by the 4th of July in many areas.

Problems associated with curlyleaf pondweed

Curlyleaf pondweed forms dense mats on the water's surface in May and June, which inhibits fishing, boating and other types of water recreation. Dense growth of curlyleaf pondweed in moving water systems can obstruct flow and can exacerbate flooding due to large amounts of biomass obstructing river channels. Dense surface mats of plant material also limit light to low-growing submersed native species and monocultures of curlyleaf pondweed often result from this competition for light. Dense vegetation at the water's surface also can stagnate the water column and inhibit oxygen exchange from the surface to the lake bottom. Decomposing plant material under the weedy canopy further reduces dissolved oxygen levels in the water column. These conditions can reduce or eliminate fish (Section 1.2) and aquatic invertebrates in dense beds of curlyleaf pondweed. Mosquitoes (Section 1.5), on the other hand, find curlyleaf pondweed beds to be the ideal habitat.

Curlyleaf pondweed typically senesces when water temperatures rise and dissolved oxygen levels begin to decline. The large amount of decomposing biomass produced from senescence releases nutrients and decreases oxygen in the water column, which further stresses the aquatic community. Algal blooms commonly occur after senescence of curlyleaf pondweed and decreased water clarity and oxygen levels can persist for the entire summer season.

Management options

Curlyleaf pondweed often requires management in order to preserve the recreational and environmental value of the bodies of water infested by the species. The most effective and efficient way to protect waterbodies from curlyleaf pondweed and other invasive aquatic species is prevention. Curlyleaf pondweed is on a number of state noxious weed lists, which make it illegal to sell or transport the species. The best way to prevent the introduction of curlyleaf pondweed into new waterbodies is to ensure that all plant material is removed from boats and trailers. Boats, trailers and gear should be thoroughly inspected, washed (with hot water) and dried before moving to a different water body to prevent the spread of curlyleaf pondweed and other invasive aquatic species.

There are a number of options for control and management in bodies of water that are already infested with curlyleaf pondweed. Physical (Section 3.4) or mechanical (Section 3.5) control options include hand removal, benthic barriers and mechanical harvesting. Hand removal by raking or hand pulling using divers can be effective tools for controlling plants in localized areas, but these efforts can be costly and time-intensive. The turion bank in the sediment should also be considered with hand removal, since regrowth from turions can quickly reinfest cleared areas. Curlyleaf pondweed can also be spread by fragments, so measures should be taken to prevent fragments and turions from spreading. Benthic barriers are also effective for curlyleaf pondweed control in localized areas. The barriers prevent regrowth from turions in the sediment and, if barriers are maintained, can provide long-term control. However, benthic barriers are labor-intensive to install and maintain and often require installation permits. Mechanical harvesting can provide temporary control of curlyleaf pondweed, but can also exacerbate the spread of fragments and turions. Management programs can include mechanical harvesting to improve boater and recreation access by effectively "mowing the lawn" to remove nuisance growth, but disposal of harvested biomass can be problematic due to the large volumes of heavy plant material. Drawdown of a body of water is an effective method for seasonal control of curlyleaf pondweed. However, drawn-down areas of shoreline can quickly be reinfested by curlyleaf pondweed plants in deeper water and by sprouting of turions in the sediment. Also, drawdowns are non-specific and will likely damage populations of desirable native submersed plants as well.

There are currently no known insect or pathogen biocontrol agents that attack curlyleaf pondweed, but sterile triploid grass carp (Section 3.6.2) can provide control of the species. However, grass carp are non-specific herbivores that will eat many native plant species. Grass carp are also illegal in many states; in states where they are allowed, their use often requires a state-issued permit and they can typically be used only in closed systems. Check with your state agency for current regulations.

Several aquatic herbicides – including diquat, endothall, flumioxazin, fluridone, penoxsulam, bispyribac and imazamox – can be used to effectively control curlyleaf pondweed. Diquat, endothall and flumioxazin are contact herbicides and are relatively fast-acting, whereas the other herbicides are systemic products that are often used as whole-lake treatments and require longer contact times for control (Section 3.7.1). Research has shown that early season treatments with herbicides can very effectively control curlyleaf pondweed and prevent turion production. Most native plant species are still dormant early in the spring, so treatment at this time prevents damage to many desirable native plants while providing selective control of curlyleaf pondweed. Since effective control early in the season prevents turion production, regrowth of curlyleaf pondweed is reduced the following year.

Summary

Curlyleaf pondweed is a problematic invasive submersed aquatic weed in the northern US and in Canada. The species grows and reproduces at very high rates and can quickly cover the entire surface of a body of water with dense monocultural growth. Dense growth of curlyleaf pondweed impedes recreation, reduces populations of native submersed plant species and alters the ecosystem so that it is inhospitable to fish and other fauna. Active management is often required to maintain the environmental and recreational value of water bodies infested with curlyleaf pondweed.

Photo and illustration credits:

Page 39: Curlyleaf pondweed infestation; Thomas Woolf, Montana Fish Wildlife and Parks Aquatic Invasive Species Bureau
Page 40 upper: Line drawing; University of Florida Center for Aquatic and Invasive Plants
Page 40 lower: Graph; Thomas Woolf, Montana Fish Wildlife and Parks Aquatic Invasive Species Bureau
Page 41: Curlyleaf pondweed; Thomas Woolf, Montana Fish Wildlife and Parks Aquatic Invasive Species Bureau

2.5 Egeria

Toni Pennington: Environmental Science Associates (ESA), Bend OR; Tpennington@esassoc.com

Egeria densa Planch.; submersed plant in the Hydrocharitaceae (frog's-bit) family
Derived from *Egeria* (Greek: water nymph) and *densa* (Latin: dense) "densely growing water plant"

Introduced from South America to the northeastern US in the 1890s
Present throughout most of the US except the upper Midwestern states

Introduction and spread

Egeria (*Egeria densa*), sometimes inappropriately referred to as *Elodea densa*, is easily confused with nonnative hydrilla (Section 2.2) and native *Elodea canadensis*. Physical similarities among the three species are responsible for the confusion in proper identification and, by extension, inconsistent naming. The popularity of egeria in home aquariums and ponds and its frequent use in biology classrooms are likely responsible for the widespread distribution of egeria across the US and elsewhere. Egeria has many common names (including anacharis and Brazilian elodea) and is commonly referred to as "oxygen weed" on many internet sites, where the species is touted for its ease of growth and ability to increase dissolved oxygen in freshwater aquariums and ponds. Many aquarists fail to consider the downsides of the plant's rapid growth rate and its effect on early-morning dissolved oxygen levels.

Plants release oxygen during the day; however, plants respire (take up oxygen) at night and cause the lowest oxygen levels to occur in the early morning. Fish kills can occur if plant density is high enough and dissolved oxygen levels become depleted overnight due to plant respiration. Like many aquatic weeds, egeria was most likely brought to the US through the aquarium trade and the species was probably first introduced to natural waterways as a result of aquarium dumping and flooding of ornamental ponds. Some states now list *Egeria densa* as a noxious weed, which may slow commercial sales and introduction to new waterbodies. The current spread of egeria is due primarily to recreational activities such as boating, fishing and the use of personal watercraft. Similar to hydrilla and other aquatic weeds, initial infestations of egeria are often found near public boat ramps, providing further evidence for this means of spread.

Description of the species

Egeria is a rooted submersed monocot that grows in a variety of fresh water bodies, including flowing and standing water. Growth of egeria is limited when the species is exposed to extremely warm (above around 90 °F) or cold (below around 40 °F) water for several weeks; however, egeria can withstand low light and low temperatures similar to Eurasian watermilfoil (Section 2.3). The species' limited tolerance for high water temperatures may explain the shift in species dominance from egeria to hydrilla during the summer in some Florida water bodies. Egeria has stems that are highly branched and can reach lengths of 25 feet or more due to the species' tolerance of very low light levels. The long stems

from a single rooted plant commonly form a canopy near the water surface that can cover an area of six feet or more, a growth habit that is observed in other canopy-forming submersed weeds. Leaves of egeria are thin, small (1-1/2 inches long and 1/8 inches wide), lance-shaped and have minute teeth along the edges that may be difficult to see without a magnifying glass. Leaves are arranged in whorls around the stem, with each composed of four to six leaves per whorl. Leaf nodes are so densely spaced at the growing tip of the plant that they are indistinguishable, but nodes are more widely spaced near the main stem and on stems lower in the water column. Branches are borne from distinct and rather predictable locations along the stems of egeria. The number of leaves per whorl doubles or even triples (up to 12 leaves per whorl) every 8 to 12 leaf nodes, which has led some to refer to these unique regions as "double nodes". These double nodes are the only location where branches and flowers are borne along the stems.

Reproduction

Egeria is dioecious, meaning that plants bear only staminate ("male") or pistillate ("female") flowers. "Female" plants (with pistillate flowers) are not known to occur outside South America. In rare cases these plants are found, but sexual reproduction and seed set are extremely rare. This has resulted in widespread distribution in the US of "male" plants (with staminate flowers) which likely have little genetic variation. Egeria spreads exclusively from vegetative propagules including stems, branches and root crowns. Branches, roots, flowers and root crowns are formed along plant stems adjacent to double leaf nodes every 8 to 12 leaf whorls. Unlike several other invasive submersed plants, egeria does not produce tubers, turions or rhizomes to facilitate spread or to provide energy storage for overwintering. Instead, egeria relies on stems and root crowns for colonization and survival during inclement conditions. Closely spaced double nodes in stem tips result in the greatest potential for growth in this region, which can make management of the species difficult. Egeria can produce a new plant from each double node along a stem fragment; this, coupled with its rapid growth rate (easily growing up to 1/2 inch per day), allows for the rapid expansion and competitive ability of the species.

Problems associated with egeria

Egeria roots in the sediment at the bottom of the water body and grows completely underwater but forms a dense mat just under the water surface. The result is a thick canopy of vegetation that spreads over large areas and impacts recreation, property values, water quality and ecosystem function.

Dense growth of egeria entangles boat propellers and impedes navigation, which often results in the unintended spread of the species when stem fragments are created after a close encounter with a boat prop. Fragments can float for days or weeks before sinking into the sediment or being stranded along shorelines. These fragments quickly form roots, which results in new colonizations or substantial increases in plant bed size that would not occur naturally. Because egeria is largely transported by human activities, infestations tend to occur near boat launches, adjacent swimming areas, marinas and boat docks. Thick mats of surface vegetation in these areas are extremely unsightly and even dangerous for users of these facilities.

Water quality may be compromised by thick surface growth of egeria. Dense growth reduces the natural mixing of water by wind and causes an increase in surface water temperature during the summer, which is harmful to fish (Section 1.2) and invertebrates. Thick mats also provide a protected growth platform for filamentous algae (Section 2.18) that are unsightly, cause odors upon decay and can support large mosquito populations (Section 1.5). Reduced wind mixing also restricts the entry of atmospheric gases (i.e., oxygen and carbon dioxide) to the water. Oxygen is necessary for fish and invertebrates, while carbon dioxide is necessary for growth of submersed plants, including algae. Similar to other invasive submersed weeds, dense growth of egeria also causes wide daily fluctuations in pH and other water quality parameters which makes infested waterways inhospitable to many aquatic animals.

Management options

Egeria has been sold as an aquarium plant in the US for more than 50 years, but it has not spread through the country as quickly as other noxious species such as hydrilla and flowering rush (Section 2.17). The first lines of defense to reduce the impacts of egeria are to prevent the introduction of the species to new water bodies and to limit its spread in waters that are already infested. The most efficient and effective preventative measure is to thoroughly remove plant fragments from boat trailers and watercraft before leaving an infested waterbody. In fact, removing all aquatic vegetation reduces the likelihood of spreading other nonnative species such as zebra mussels and other inconspicuous species such as New Zealand mudsnails. The cost of prevention (e.g., through signage, boat inspections, boat washing stations, etc.) is orders of magnitude less than the cost of managing existing populations because once egeria is established it is extremely difficult, and most would argue impossible, to eradicate.

Physical (Section 3.4) and mechanical (Section 3.5) controls for egeria are similar to those for other submersed weeds, largely due to their ability to establish new colonies from stem fragments. As a result, the benefits and drawbacks of various control methods are similar among the species. Hand removal and the use of benthic barriers can be selective; however, these methods are very laborious and time-intensive. Because egeria does not produce tubers or turions, the likelihood of reinfestation after benthic barriers are removed or when hand pulling is completed is reduced, provided both methods are employed with vigilance. Mechanical harvesters can clear large areas for boat navigation; however, harvesters can produce thousands of fragments that can expand the population. Since harvesters essentially mow the upper portions of the plant, the need to remove stem tips after mechanical harvesting cannot be understated; otherwise, stem tips float away and spread the plant to new habitats within a water body. In addition, multiple harvests are usually required during the peak growing season due to the rapid growth rate of egeria.

Water level drawdowns may be used where feasible to control egeria in regulated water bodies (e.g., irrigation canals and reservoirs for power generation or flood control). Egeria may be the submersed aquatic weed most susceptible to drawdown and desiccation because seeds, tubers or turions are not produced to allow for re-growth; however, plant fragments within mounds that maintain adequate moisture are known to be viable after as much as 30 days of dewatered conditions. Plants are particularly vulnerable during winter drawdowns when dry and freezing conditions are present.

The required duration of dewatering depends on various climatic and sediment conditions such as relative humidity, temperature and sediment density (the ability of soil to retain water). Disadvantages to drawdown include lack of specificity (nontarget native plants and wildlife are impacted) and loss of the water for other purposes such as hydropower, irrigation and recreation.

Currently, the only biocontrol agent available in the US for reducing egeria biomass is the sterile grass carp (Section 3.6.2). Grass carp have been stocked following drawdown in some locations, which has led to long-term control. Sterile grass carp effectively control egeria in areas where low water temperature does not limit their feeding; unfortunately, egeria is capable of positive and sustained growth in climates cooler than those required for active grass carp feeding, so effectiveness may be limited under those conditions. Research in California on a leaf-mining fly (*Hydrellia egeriae*) concluded that the host range of *H. egeriae* is too broad for use in the US as it indiscriminately oviposits on both egeria and the native *Elodea canadensis*.

Herbicides commonly used to control egeria include the systemic herbicides fluridone and penoxsulam and the contact herbicides copper, endothall (mono(N,N-dimethylalkylamine salt) and diquat (Section 3.7.1). The list of herbicides that can be used to effectively control egeria is very limited compared to those used to control Eurasian watermilfoil. Egeria is a monocot and is therefore not susceptible to 2,4–D or triclopyr. Egeria is often found in systems with flowing water, which makes the use of slow-acting systemic herbicides challenging because plants require a long exposure time in order for systemic herbicides to provide effective control. The growth of egeria in flowing water systems coupled with a limited number of effective herbicides make egeria a difficult plant to control with herbicides.

Summary

The popularity of egeria in the aquarium trade and in biology classrooms has substantially contributed to its widespread distribution in the US, Europe, Asia, New Zealand, Japan, Chile, Mexico, Canada and Australia. The spread of egeria between water bodies is largely due to trailered boats and other watercraft that transport fragments. Long-lived stem fragments are easily spread by currents and watercraft within infested water bodies. When these fragments come into contact with sediments on the lake bottom or the margins of the water, the fragments form roots, plantlets develop and new colonies of egeria rapidly become established. Egeria tolerates a wide range of water quality characteristics, sediment nutrient levels and light levels and commonly grows in similar habitats favorable to Eurasian watermilfoil. As a result, it is likely that egeria can invade and colonize areas that currently support growth of Eurasian watermilfoil.

Photo and illustration credits:
Page 43: Egeria infestation; Toni Pennington, ESA
Page 44 upper: Line drawing; University of Florida Center for Aquatic and Invasive Plants
Page 44 lower: Egeria; Toni Pennington, ESA
Page 45: Egeria; Toni Pennington, ESA

2.6 Fanwort and Cabomba

Brett W. Bultemeier: University of Florida, Gainesville FL; bwbult@ufl.edu

Cabomba caroliniana A. Gray; submersed plant in the Cabombaceae (watershield) family. Derived from *Cabomba* (an aboriginal name per botanist Asa Gray in 1848) and *caroliniana* (having a range that includes North and South Carolina in the US)

Native to the southern US, although some "populations" appear introduced from the aquarium industry. Found in the southeast, northeast, midwest and Pacific northwest US, Australia and New Zealand

Introduction and spread

Fanwort (*Cabomba caroliniana*) is one of five species of the genus *Cabomba* and the only one broadly distributed in the United States (although *C. haynesii* and *C. palaeformis*, both known by the common name fishgrass, reportedly occur in Miami-Dade County in extreme southern Florida). There are three varieties of fanwort, but only two (*C. caroliniana* var. *caroliniana* and var. *pulcherrima*) are considered native to the US. In addition to native populations of

fanwort found throughout most of the eastern US, there is also a new type of fanwort that was likely introduced via the aquarium trade. Members of the genus *Cabomba* appear very similar to one another and are difficult to identify with certainty; even plant taxonomists are currently unable to clearly define species and subspecies of *Cabomba*. However, it is clear that many of the new populations of *Cabomba* found throughout the midwestern US and Canada are invasive and have other characteristics that distinguish them from native populations. These invasive types will hereafter be referred to as "green cabomba"; the term "fanwort" will refer to members of the species *C. caroliniana*.

There is little information outlining the introduction of green cabomba, but research in the early 1980s revealed that the aquarium trade had discovered or developed a variety of fanwort that was solid (or nearly solid) green. Populations of green cabomba began to appear and rapidly expand in the midwestern and northwestern US, Canada and Australia in the early 1990s. Because these populations are similar in appearance and invasiveness, it seems likely that they were introduced from a common source – probably the aquarium trade. In addition to these new invasions of green cabomba, invasive behavior has also increased in native populations of fanwort in the southeastern and northeastern US. Cabomba populations in Australia and New Zealand are considered invasive and attempts are being made to eradicate the species.

Description of the species

Fanwort is a perennial dicotyledonous plant that roots in the sediment and grows entirely submersed in the water column. It colonizes new areas through prolific root growth or through shoot fragments that become rooted in the sediment. The species typically grows in shallow waters, but can be found at depths of up to 30 feet if the water is clear. Abundant branching occurs at the root crowns and base of the plant. Shoots grow to the surface of the water and continue to elongate, producing thick mat-forming canopies. Stems are round to slightly compressed and range in color from green to red (although stems are always green in green cabomba). Submersed leaves are opposite, fan-shaped, finely divided with as many as 200 terminal points on a single leaf and range from green to red. Leaves can vary greatly in size, but leaves near the tip of the plant are usually smaller and closer together than lower leaves.

Flowering occurs on the surface of the water on branches with floating leaves. Floating leaves look very different from submersed leaves and are alternate, smooth and linear-elliptic to ovate. Flowering stems bear single bisexual white flowers with 3 petal-like sepals and 3 petals; some flowers have yellow spots or purplish margins. Populations of native fanwort flower profusely, but green cabomba produces few flowers.

Fanwort prefers to grow in acidic water with a pH of 4 to 6 and growth is inhibited when water pH is above 7. Green cabomba, however, can survive in water with a higher pH and growth is not affected unless pH is 8 or higher. Fanwort is considered a more tropical species and proliferates in the southeastern US, whereas invasive green cabomba has colonized the much colder climates of the midwestern US and Canada and has adapted to overwinter there. During late fall when temperatures begin to drop, green cabomba stems break off and turion-like structures form at the apical tip. When warmer temperatures return in early spring, these fragments will begin to elongate and form adventitious roots.

Variations in color are the most significant barrier to separating members of the genus *Cabomba*. Most descriptions of fanwort list color as ranging from green to red, with red coloration most common in warmer temperatures and green in cooler temperatures. True fanworts – for our purposes, *Cabomba caroliniana* – do often have green leaves close to the base of the plant and red to purple leaves near the tip of the plant, but this is highly variable. Some populations may be entirely red to purple with no green (these plants are most likely *C. caroliniana* var. *pulcherrima*); however, other plants may appear red to purple but have green leaves in deeper water. In contrast, green cabomba is always entirely green and

water temperature has no effect on color. These color differences provide evidence to support the theory that green cabomba is unique from native fanworts and there are differences in physiological responses as well. For example, research has shown that green cabomba grows more quickly, tolerates cold temperatures, survives under a wider range of water pHs and may be more tolerant of some herbicides than native fanworts.

Reproduction

Fanwort and green cabomba reproduce using multiple strategies. Both spread via vegetative fragmentation; a single leaf node can produce roots and grow into a new plant. As such, contaminated watercrafts, trailers and live wells can transfer these species to new areas. Also, both species grow in slow flowing canals and rivers, so plant fragments can travel long distances on currents until they settle in a suitable habitat. Fanwort spreads primarily through vegetative fragmentation, but sexual reproduction does occur. Flowers are usually pollinated by insects, although self-pollination can occur as a result of wave action. Flowering is a two-day event; flowers emerge and can be pollinated on the first day and are closed and pulled below the surface of the water for seed formation on the second day. Seed viability is very low in fanwort and whether viable seed production occurs in green cabomba is unknown.

Problems associated with fanwort and cabomba

Species of *Cabomba* produce mat-forming canopies that can become quite dense, particularly when these mats are produced by green cabomba. Dense canopies decrease light penetration through the water column, which can displace or eliminate other desirable or native plant species (Section 1.1), thus creating a monoculture of fanwort or green cabomba. This lack of diversity can impact fisheries (Section 1.2) and waterfowl (Section 1.3), especially when coupled with the reduced dissolved oxygen levels that result from poor penetration of oxygen through dense vegetation. These thick mats can also impede navigation and recreational use of the water body and can have negative economic impacts on the industries that utilize these resources. Plant fragments may also clog drainage pipes, canals, intakes, pumps and other structures, which can impede irrigation, drainage and flood control efforts.

Management options

Mechanical control (Section 3.5) is unlikely to be successful in eradicating fanwort and green cabomba from an aquatic system, since harvesting can produce fragments that can root, form new plants and quickly recolonize the water body. Also, extensive root systems are often undisturbed by harvesting and new plant growth from these roots can quickly re-infest an area after harvesting operations are concluded. Drawdowns (Section 3.4) can be used to control fanwort and green cabomba, but are not practical in areas where the waters are heavily utilized for recreational activities and elimination of the resource is not an option. Because fanwort grows best in low-pH water and growth is inhibited at higher pH, it may be possible to use lime to increase pH as a control strategy. Fisheries biologists have added agricultural lime to farm ponds and noted control of fanwort and improved fish production due to the resulting increased pH. There are no known biological control agents (Section 3.6) for fanwort, although the generalist herbivore grass carp (Section 3.6.2) will provide some control of the species. Because green cabomba was probably created by the aquarium industry, it is unlikely that biocontrol agents will be identified for green cabomba, since these agents would also likely feed on native fanworts.

Chemical control (Section 3.7.1) of fanwort is possible with several herbicides, but control of green cabomba is much more challenging. Contact herbicides such as diquat, endothall (amine salt) and flumioxazin, along with the systemic herbicide fluridone, can be used to control fanwort. However, diquat and fluridone have little effect on green cabomba.

Flumioxazin is reportedly effective on green cabomba and high rates of the amine salt of endothall can also reduce biomass, but toxicity to fish is a concern when using high rates of endothall amine. Thus, options for chemical control of *Cabomba* species – particularly green cabomba – is limited at this time.

Summary

Although fanwort is a native species, populations of green cabomba behave like – and have impacts similar to – an invasive species. Native populations of fanwort are prevalent in the southeastern US, whereas green cabomba is more common in Canada and in the midwestern, northeastern and northwestern US. Identifying species in the genus *Cabomba* is challenging, which makes it difficult to characterize invasions by green cabomba; however, it is clear that its rapid spread to new areas of the US over the last few decades is troubling. Furthermore, the rapid spread of green cabomba through fragmentation and a lack of available management tools is cause for concern since it may be difficult to limit the further spread and impact of this plant throughout the US.

Photo and illustration credits:

Page 47: Cabomba infestation; Brett Bultemeier, University of Florida
Page 48 upper: Line drawing; University of Florida Center for Aquatic and Invasive Plants
Page 48 lower: Color variation in cabomba/fanwort; Brett Bultemeier, University of Florida
Page 49: Cabomba flower; Lyn Gettys, University of Florida
Page 50: Cabomba population; Lyn Gettys, University of Florida

2.7 Starry Stonewort

Tyler D. Geer: Clemson University, Clemson SC; tgeer10@gmail.com

Nitellopsis obtusa (Desvaux in Loiseleur) J. Groves, submerged macro-alga in the Characeae family

Native to Europe and Asia, introduced to North America in the St. Lawrence River, presumably in ballast water of trans-oceanic ships. Currently found in inland aquatic ecosystems in the midwest and northeast US and Ontario, Canada

Introduction and spread

Nitellopsis obtusa (starry stonewort) is an invasive green macro-alga native to Europe and Asia and records indicate that it was introduced to North America in the St. Lawrence River in the 1970s. It is likely that starry stonewort was transported to the Great Lakes in North America in the ballast water of a trans-oceanic shipping vessel. There was an apparent lag in expansion of starry stonewort until the 2000s, several decades after its introduction. This may be due in part to a lack of awareness or systematic search efforts, coupled with the difficulty of identifying this alga and its ability to initially "hide" among other algae and vascular plants. In 2005, starry stonewort was reported from Upper Little York Lake in New York and had expanded into Pennsylvania, Indiana and interior Michigan by 2012. The first population of starry stonewort was identified in Wisconsin in 2014, followed by the discovery of populations in Minnesota and Vermont in 2015. Star-shaped bulbils of starry stonewort were found in Lake Simcoe (Ontario) in 2009 and the species was reported in Presqu'ile Bay (Lake Ontario) and Lake Scugog (Ontario) in 2016. At the time of this writing, starry stonewort is present in North America in Ontario, Canada and seven Great Lakes states in the northern US.

Almost all spread of starry stonewort is attributed to human-mediated transport on boats and boating equipment, indicated primarily by the preponderance of new infestations around watercraft access points such as boat ramps. For example, a study of lakes in New York revealed that starry stonewort was not present in 20 lakes that lacked boat launches, even though those lakes were within the most heavily starry stonewort-invaded region of New York. It is unlikely that starry stonewort has reached the full extent of its potential range in North America and several attempts have been made to predict regions of the US that could be susceptible to new invasions. One study that used a climate-based ecological niche model predicted that large portions of the mid-Atlantic, intermountain

West and Great Plains regions of North America could provide habitat for starry stonewort, although the species has not yet been reported in these regions. Likewise, a water chemistry-based model identified areas of the northeastern US, including eastern New York and western Vermont, as suitable regions. There is no reason to expect that starry stonewort will not colonize much of the eastern and midwestern states of the US.

Description of the species

Starry stonewort is a robust Characean macro-alga and is a relative of the common native algae genera *Chara*, *Nitella* and *Tolypella*. In its introduced range, starry stonewort grows in a manner similar to rooted, submersed perennial macrophytes. The main "stem" or thallus (plural: thalli) of starry stonewort is slender (up to 1/12 inch in diameter) and emerges from the sediment, where it is anchored by a network of rhizoids (colorless root-like structures). Thalli have branchlets that are arranged in whorls of 5 to 8 around nodes, which can give rise to further branchlets. Starry stonewort forms dense, monotypic stands with a disorganized appearance and will grow to just below the water's surface.

New growth of starry stonewort is easily confused with its Characean relatives, especially species of *Chara*, and starry stonewort is often misidentified as "super-chara". However, starry stonewort is smooth to the touch, in contrast to the rough or crunchy texture of *Chara*; it has no odor, whereas *Chara* often smells like garlic or onion; and starry stonewort produces bulbils (clonal seed-like reproductive structures), while *Chara* does not. Starry stonewort's common name is derived from its unique star-shaped bulbils, which can be found attached to rhizoids in the sediments around populations of starry stonewort.

Reproduction

In its native range of Europe and Asia, starry stonewort reproduces using both sexual (oospores) and asexual/vegetative (bulbils and fragmentation) means. However, only male reproductive structures have been found to date in North American populations of starry stonewort, which results in reproduction entirely by vegetative means in the US. New growth of the species emerges from sediment in mid- to late spring; peak biomass typically occurs between late June and late August, depending on latitude, and starry stonewort can grow until November in some regions. Bulbils appear to be most prevalent in early spring (when the alga is starting to emerge from the sediment) and in the late summer and fall (prior to the onset of winter and accompanied by natural senescence of the population). Bulbils seem to be short-lived but likely remain viable for at least 6 months.

Problems associated with starry stonewort

Starry stonewort grows entirely underwater as a submersed alga and its growth potential appears limited primarily by wave action and the depth of light penetration. Starry stonewort is commonly found at depths of 5 to 15 feet but has been found in water as deep as 30 feet. This ability to colonize water that is deeper than most native submersed species can tolerate may allow it to obtain a foothold before spreading into the shallower margins of lakes where most native plants colonize. Although starry stonewort is an alga, dense populations can cause problems similar to those associated with invasive aquatic plants, such as reducing light and oxygen availability and interfering with human uses of infested waters. Given starry stonewort's capability for vigorous growth and rapid spread, in many aquatic ecosystems it may only take a few years for a seemingly benign colony of the species to become a wide-spread and problematic infestation.

Management options

Starry stonewort can be controlled relatively easily, but requires persistence to be effective. The initial step is to identify the level of infestation and to determine the management objectives (e.g., extirpation or local eradication from a water resource, maintaining access for boats, habitat improvement, protection of nearby uninvaded waters). Most starry stonewort management programs focus on two components: 1) prevention of off-site movement and reintroductions of starry stonewort propagules, and 2) control of populations within infested water resources. The specific management approach depends on the degree of invasion and the maturity of the population(s) and can be broadly grouped into four scenarios: 1) starry stonewort is absent; 2) incipient populations are present, 3) starry stonewort populations are established, and 4) widespread, mature starry stonewort populations are present.

If starry stonewort is absent, the focus is on preventing the introduction of fragments and bulbils from contaminated boat trailers and watercraft. This can be achieved by thorough inspections of boat trailers and watercraft before leaving an infested water resource, along with more intense cleaning such as pressure washing. It is important that prevention actions are coupled with routine and strategic monitoring of vulnerable areas of aquatic ecosystems. Most new infestations go unnoticed until populations reach the water's surface, so monitoring vulnerable areas (particularly

accesses or physical linkages with infested waters) can improve the chance that new infestations are quickly detected. Early detection improves the effectiveness of rapid responses to incipient starry stonewort populations (e.g., newly discovered populations in less than 5 acres), reducing management costs over the long term. Extirpation of starry stonewort may be possible at this stage and control tactics should be implemented early in the growing season when the algae are less than 1 foot tall. Physical control tactics (Section 3.4) such as hand removal and benthic barriers can be employed and mechanical harvesting (Section 3.5) can be used if the harvesting apparatus can reach the depth at which starry stonewort is growing. Algaecide applications (Section 3.7.1) provide the most effective control of starry stonewort and can be used alone or as a follow-up treatment after physical or mechanical harvesting. Copper-based algaecides chelated with ethanolamine, used alone or in combination with endothall and/or adjuvants (Section 3.7.3), can provide effective control of starry stonewort. Flumioxazin may also be useful, although use of a herbicide to control starry stonewort may also affect nearby nontarget aquatic plants. Algaecides should be applied directly to starry stonewort, so new infestations or early season growth at the bottom of a system should be targeted with trailing hoses or injection pipes (Section 3.7.4).

Management of established populations (e.g., algae reaches the surface of the water, populations are greater than 5 acres, multiple populations are present) requires aggressive action to restore and maintain the water resource. As with incipient infestations, control tactics should begin as early in the growing season as possible (algae less than 2 feet tall), and mechanical and chemical control tactics are most applicable at this scale. If management goals are extirpation or whole-season maintenance of starry stonewort to allow boat access, multiple algaecide applications or harvesting efforts will be necessary to achieve “successful” control. Dense stands that develop throughout the growing season may require algaecide applications to the surface of the water, as dense growth may impede trailing hoses and prevent mixing of algaecide in the water column. There are currently no biological control agents (Section 3.6) are available for starry stonewort. Grass carp (Section 3.6.2) may be useful for biological control of starry stonewort but reports regarding their efficacy are lacking.

Control measures for widespread, mature populations of starry stonewort are similar to those for established populations, but management efforts may be limited by resources. For density dependent tactics such as algaecide treatments, where costs depend on the quantity of starry stonewort targeted for control, treatments should be used early in the growing season so they can be distributed over a greater area. If repeated treatments are not possible, management efforts should be timed to ensure that starry stonewort is controlled during a relevant economic, ecological, or socio-political time period. For example, if boat navigation is the most important management objective, treatments can be timed to control starry stonewort when boat traffic is the greatest, such as around summer and fall holidays. As for established starry stonewort populations, the most effective tactics for control of widespread, mature starry stonewort populations are mechanical harvesting and algaecides. The key to managing dense, mature populations of starry stonewort is persistence. Multiple algaecide applications, or mechanical harvesting of the upper portion of the population followed by an algaecide application to control the remainder, can overcome biomass limitations and restore the intended uses of a water resource.

Summary

Starry stonewort is a relatively new invasive aquatic alga in North America that has spread rapidly among inland aquatic ecosystems in the Great Lakes and Northeast regions of the US despite its inability to reproduce using sexual means. Overland dispersal is attributed to watercraft and associated equipment, making inspection and control of “hitchhikers” the first line of defense against its spread. Bulbils and fragments that escape or survive prevention efforts can colonize new aquatic ecosystems and result in new populations that have substantial ecological, economic, and socio-political impacts in invaded and connected water resources. If detected early in its colonization, it may be possible to extirpate starry stonewort from an aquatic system. If starry stonewort is allowed to persist in a water resource as a result of failure to detect the infestation or failure to initiate action early, significant resources and persistent use of control tactics will be needed to control the alga, maintain use of the water resource and prevent infestation of nearby or connected waters.

Photo and illustration credits:

Page 51 upper: Starry stonewort infestation; Tyler Geer, Clemson University
Page 51 lower: Starry stonewort on boat propeller; Tyler Geer, Clemson University
Page 52 upper: Starry stonewort line drawing; from “A decade of starry stonewort in Michigan” by G. Douglas Pullman and Gary Crawford (Summer 2010 Lakeline 36-42), used with permission
Page 52 lower: Starry stonewort bulbils attached to rhizoids; Steve McComas, Blue Water Science, St. Paul MN, used with permission
Page 53: Harvester (Huron Lakes Weed Control, Pickney MI); Tyler Geer, Clemson University

2.8 Parrotfeather

Ryan M. Wersal; Minnesota State University Mankato, Mankato MN; ryan.wersal@mnsu.edu

Myriophyllum aquaticum (Vell.) Verdc is a heterophyllous plant (distinct submersed and emergent leaves on the same plant) in the Haloragaceae (watermilfoil) family
Derived from myrios (Greek: *numberless*), phyllon (Greek: *leaf*) and aquat (Latin: *water*)
"water plant with many leaf divisions"

Introduced from South America to New Jersey in the 1890s
Present throughout the southern US; north to New York; west to Washington state and Idaho

Introduction and spread

Parrotfeather is native to South America and is a member of the Haloragaceae family. The species is closely related to the invasive species Eurasian watermilfoil (Section 2.3) and has been introduced to Southeast Asia, Australia, New Zealand, Japan, South Africa and North America. The earliest specimen recorded in the US was collected in Haddonfield, New Jersey in 1890. Parrotfeather is widespread in the United States, infesting nearly all southern states, Hawaii, and as far north as New York on the east coast and Washington on the west coast.

Parrotfeather is a rooted plant that grows well in the moist soils of shallow wetlands, slow moving streams, irrigation reservoirs, canals, edges of lakes, ponds, sloughs and backwaters. The species tolerates frequent inundation of salt water as long as concentrations remain below 4 parts per thousand and can grow in water depths of up to 10 feet if the water is clear and light penetration is sufficient to support growth and colonization. Once stems reach the surface of the water, the creeping growth habit of parrotfeather can quickly cover large expanses of the water surface. Parrotfeather is not seriously affected by frost; however, a hard freeze may kill emergent shoots in northern latitudes. The species overwinters in submersed form and resumes growth when water temperatures reach 45 °F.

Because parrotfeather is widely adapted, it is frequently used as an ornamental plant in ponds and water gardens. In addition, the species is sold as an "oxygenating" plant for aquariums across the US. The spread of parrotfeather is almost exclusively attributed to humans. For example, in the 1980s aquarium plant growers in the San Francisco Bay area planted parrotfeather in local waterways to have a convenient source of plant material to sell to their customers. Parrotfeather's ease of cultivation and attractiveness as a pond plant are likely the most common means of spread of the species and have aided in its escape and subsequent colonization of natural areas.

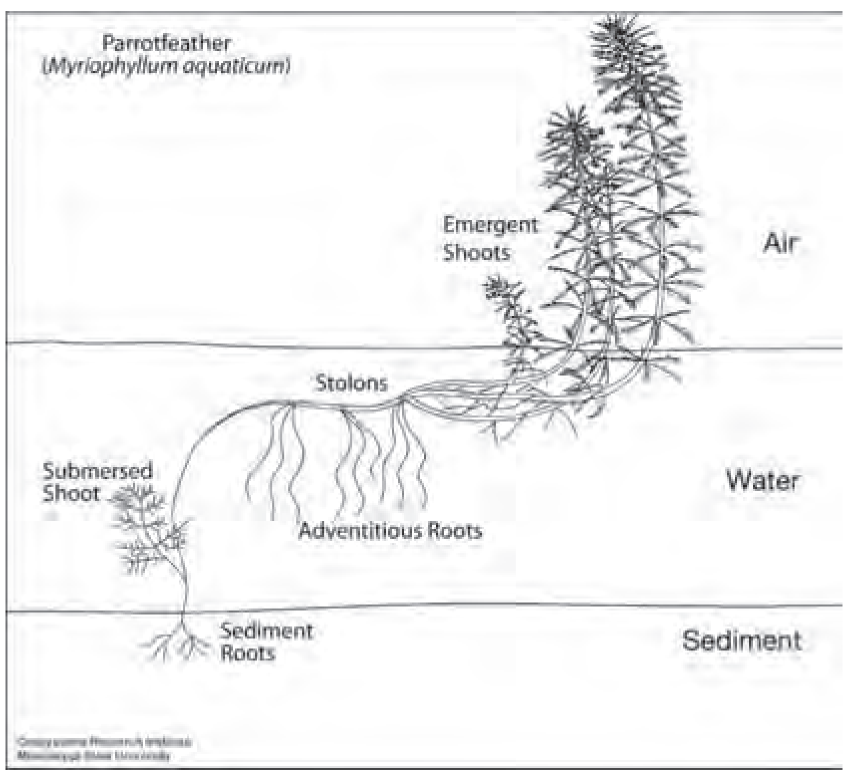
Little information exists regarding the spread of parrotfeather by animals. The species does not produce seed, turions or tubers and the leaves are generally unpalatable to most animals. However, parrotfeather is tolerant of dry conditions and may be spread if cattle or other rangeland animals utilize ponds and wetlands infested with the species because fragments may become entangled in fur or lodged in hooves and transferred to other areas. Waterfowl (Section 1.3) or migratory birds may also aid in spreading parrotfeather if fragments are transported in feathers, although there is no direct evidence of this mode of transport.

Description of the species

Parrotfeather is an evergreen stolon-forming perennial with sturdy stems that have submersed and emergent portions. The emergent and submersed leaves of parrotfeather differ in appearance from one another, a phenomenon known as heterophylly. Emergent leaves are whorled, stiff, grayish green, feather-like and usually have 20 or more divisions on each leaf. Leaves borne on submersed shoots are reddish orange, thread-like, pectinate and arranged in whorls of four to six at each node. Submersed shoots grow mostly in a vertical manner until they reach the water surface, when growth of the plant changes to the emergent form. Stems initially creep horizontally along the surface of the water with extensive branching at each node; the horizontal growth is followed by vertical growth of new stems. Parrotfeather also produces adventitious roots at each node that are important for nutrient uptake and mat formation since roots from different plants often become entangled. Flowering typically occurs from March to May in the leaf axils of emergent shoots. Parrotfeather is dioecious [plants produce either pistillate ("female") or staminate ("male") flowers], but plants with staminate flowers are not found outside South America and are rare even in native populations of South America. Because seed set requires the presence of both types of flowers, seed production is not known to occur in parrotfeather and the species reproduces exclusively by vegetative means.

Reproduction

Parrotfeather lacks specialized structures such as seeds, tubers, turions and winter buds that often facilitate overwintering in many aquatic plants. Instead, plants survive adverse conditions by switching between the submersed and emergent growth forms. The switching of growth forms is accomplished by reallocation of resources (carbohydrates) throughout the growing season. The majority of starch is stored in the stolons of parrotfeather, making it readily available to both the submersed and emergent leaves.



The submersed form can overwinter in colder water where the emergent form would be killed. Additionally, the emergent form is somewhat tolerant to desiccation and can therefore survive for a period of time in areas that have been de-watered as long as the soil remains moist. The species reproduces solely through the fragmentation of emergent and submersed shoots. Both types of shoots are easily fragmented; once fragmentation occurs, adventitious roots are rapidly formed, which allows the plant to anchor in the sediment and take up nutrients needed for growth.

Problems associated with parrotfeather

Little information exists regarding the direct impact of parrotfeather infestations on fish and wildlife populations. Dense beds of parrotfeather reduce dissolved oxygen in the water column, which may be detrimental to fish populations. Parrotfeather may outcompete more desirable aquatic plant species [e.g., pondweeds (*Potamogeton* sp.)

and coontail (*Ceratophyllum demersum*)] which are readily utilized as a food source by waterfowl. Dense surface mats of emergent growth shade out and eliminate native submersed plants, which also reduces populations of macroinvertebrates that are important to waterfowl as well as for fish (Section 1.2).

The creeping growth of parrotfeather can quickly cover large areas of the water surface, which can impede navigation, stream flow and runoff and can increase the duration and intensity of flooding. Dense growth of parrotfeather in irrigation canals of the western US have limited the water supply needed for crop irrigation. Also, the species infests major river systems and tributaries in a number of developing countries and poses a direct threat to potable water supplies. In addition, dense infestations of parrotfeather offer mosquito adults, eggs and larvae a refuge from predation, which may lead to increases in mosquito-borne diseases that affect wildlife and humans (Section 1.5).

Parrotfeather competition

Parrotfeather is generally not a strong competitor, especially as species richness increases, but it can cause problems in small ponds and ditches where disturbances are frequent. In fact, disturbance is often cited as a primary means for dispersal of parrotfeather, especially when mechanical control (Section 3.5) is utilized. Because parrotfeather relies on fragments for growth, it can re-colonize after a disturbance more quickly than species that must regrow from seeds, tubers or turions. Other aquatic plants with a creeping growth habit (such as alligatorweed) can overtake infestations of parrotfeather and reduce its growth.

Management options

Parrotfeather is often overlooked until it reaches nuisance levels, which allows the species time to become established and expand its range. Although parrotfeather is not considered a widespread nuisance, control of the species after it has invaded an area is difficult regardless of the method used. The most effective means to avoid infestations is to prohibit the sale of parrotfeather by the water garden and aquaculture industries and to ensure plant fragments don't hitchhike on boat trailers, as these are the primary means of spread in the US.

Several methods – including chemical (Section 3.7.1), mechanical and biological control (Section 3.6) – have been evaluated with mixed results. Chemical and mechanical methods can provide short to medium term control of parrotfeather. Herbicides are used most often and results are dependent upon herbicide choice. The use of mechanical methods has received less attention, but their use may encourage fragmentation, regrowth and further spread of parrotfeather. There are currently no effective biological control agents available in the US.

Foliar applications of herbicides – including 2,4-D, imazapyr and triclopyr – have resulted in consistent control of parrotfeather. Glyphosate and diquat are generally not recommended because only emergent shoots are killed and plants often regrow at greater densities. Foliar applications of carfentrazone-ethyl and flumioxazin will not control parrotfeather in the long term; however, these herbicides can be combined with 2,4-D to provide excellent control of small infestations of parrotfeather. The effectiveness of herbicides applied to the water column has not been studied as extensively as foliar applications. However, of those herbicides tested as submersed treatments, only triclopyr and the butoxyethyl ester formulation of 2,4-D have provided good control of parrotfeather. The effectiveness of herbicide treatments is site-specific and is influenced by environmental conditions at the time of application. In addition, multiple applications are often necessary to completely control parrotfeather. Overall, the auxin class of herbicides usually offers the best results when controlling parrotfeather.

Mechanical harvesting methods such as raking, chaining (long chains of sharp blades pulled by tractors) or hand pulling can provide temporary control of small infestations of parrotfeather. However, these approaches are very labor intensive as dense mats are heavy and difficult to remove from the water. Also, harvested plants have little utility as a mulch or animal forage and must be taken to a landfill. Parrotfeather is often tolerant of mechanical disturbance; in fact, the repeated use of mechanical control methods actually favors the formation of new infestations of parrotfeather because numerous fragments are produced during harvesting operations. As a result, care must be taken to remove all plant parts (emergent shoots, submersed shoots, root crowns and fragments) after mechanical harvesting to reduce the risk of re-infestation.

Cultural methods (Section 3.4) such as drawdown and dredging might provide effective control of parrotfeather in some areas, provided the drawdown is maintained long enough to completely dry the soil since parrotfeather can survive in moist soil. Winter drawdowns in the southern US would likely not be effective because the rainfall that occurs during this time is usually sufficient to keep soil moist enough for parrotfeather survival. Dense mats of parrotfeather also serve as an insulator against cold weather and water loss, which keeps sediments and root crowns moist and viable. Winter drawdowns in the northern US may be effective if the upper 6 to 8 inches of the soil freezes and causes death of the root crown. Summer drawdowns can provide a measure of control if sediments dry to the point of cracking, which should dry plant mats and root crowns and result in plant death. However, if a large amount of biomass is present it may prolong the drying process by trapping moisture underneath the thick mat. The moisture will be enough to keep parrotfeather alive until more favorable growing conditions return. Dredging can be used to control parrotfeather infestations, but the technique is expensive and control is short-lived because it is difficult to ensure removal of all plant fragments and root crowns.

Summary

Since its introduction in 1890, parrotfeather has spread down the East Coast, across the southern US and along the West Coast as far north as Washington and Idaho. Having both submersed and emergent growth forms allows parrotfeather to invade a wide range of habitats and may give this species an advantage over other aquatic plants, especially during times of adverse environmental conditions such as droughts or floods. Although parrotfeather does not share the rapid invasion characteristics associated with other noxious species such as Eurasian watermilfoil and hydrilla, it can quickly colonize and overtake a variety of habitats in a short period of time. Parrotfeather is often overlooked until it becomes firmly established; once this occurs, parrotfeather has shown great resiliency towards many of the current management techniques. Parrotfeather is not included in the Federal Noxious Weed list. As a result, buying, selling and transporting this species is not restricted in most states. Parrotfeather is widely sold in the water garden industry and is one of the most popular plants sold for this purpose. The continued sale and transportation of this species is responsible for most of its spread and will further exacerbate future nuisance problems associated with parrotfeather.

Photo and illustration credits

Page 55: Emergent growth form of parrotfeather. Ryan Wersal, Minnesota State University Mankato
Page 56 upper: Flowers in the leaf axils of parrotfeather. Ryan Wersal, Minnesota State University Mankato
Page 56 lower: Line drawing of parrotfeather growth. Geosystems Research Institute, Mississippi State University
Page 57: Population of parrotfeather (note reddish-orange leaves on submersed form). Ryan Wersal, Minnesota State University Mankato

2.9 Floatinghearts

Ian J. Markovich, Joseph W. Sigmon and Lyn A. Gettys: University of Florida FLREC, Davie FL; ijmarkovich@ufl.edu, jsigmon@ufl.edu and lgettys@ufl.edu

Floatinghearts (*Nymphoides* sp.) are rooted, floating-leaved plants in the Menyanthaceae (buckbean) family. Derived from *nymph* (Latin: water), *oides* (Greek: resembling, similar); "*Nymphaea-like*"

Introduction and spread

There are around 50 species of floatinghearts and seven have been found in North America. Two of these species (bananalily and little floatingheart) are native to North America, and three species (yellow floatingheart, snowflake lily and crested floatingheart) were introduced through the ornamental water garden market and have escaped cultivation.

The introduction pathways of the remaining two species (Gray's floatingheart and Humboldt's floatingheart) are unclear, but it is likely they are also water garden escapees or have spread due to "natural" range expansion, which was possibly facilitated by flood events or transportation by animals.

Although there are native species of *Nymphoides* in North America, some of the introduced species have had a significant impact on natural environments. For example, crested floatingheart has jeopardized south Florida's flood control (canal) systems by obstructing water flow and choking out waterways while displacing native species. Resource managers in South Carolina also struggle to manage introduced floatinghearts in their reservoirs and lakes. In addition to these direct and immediate threats to the ecosystem, hybridization between native species (bananalily and little floatingheart) and between introduced and native species (crested floatingheart and bananalily) has been documented in South Carolina, which could change the native gene pool and impact management options.

Description of species

Nymphoides is a remarkably diverse genus, but member species share some similarities. All exhibit nymphaeid growth form, meaning they are rooted in the substrate and produce leaves and flowers that float on the surface of the water. These cosmopolitan species differ from one another in growth habitat, flower appearance, structure, and reproductive strategy. Molecular work and ancestral reconstruction has shown that this genus has evolved independently at least twice through time. All of the seven species of floatingheart that have been reported in North American water bodies have multiple long petioles (leaf stalks) that each bear a single leaf. Most of the seven species reproduce through vegetative means, although some produce viable seeds as well.

Native floatinghearts

Bananalily (*N. aquatica*) and **little floatingheart** (*N. cordata*) are native to North America. Bananalily is found throughout the eastern and central United States. The leaves are oval to kidney-shaped, with a dark green upper surface and a purple corky underside that often has prominent veins. Flowers range from 1/3 to 3/4 inch wide and have five white, smooth petals with a papery margin and a yellow center. Little floatingheart is found throughout the eastern United States and Canada and has heart-shaped to oval leaves that are often variegated with a reddish-purple upper surface and an underside that is mostly smooth (like fine sandpaper). At only 1/5 to 2/5 inch wide, the flowers of little floatingheart are smaller than those of bananalily, but are otherwise similar in appearance. The primary reproduction method for both species is the production of "ramets", which are rhizome-like clusters that form at the base of the leaf. These ramets break off and float away to become a new plant.

Introduced floatinghearts

Crested floatingheart (*N. cristata*) is native to southeastern Asia. Leaves are up to 8 inches long, 6 inches wide and heart-shaped; the upper surface of the leaf is green, usually with a red margin, and the underside is smooth and reddish. This species produces white five-petaled flowers that are up to 1 inch across and have a membranous margin and a crest or ridge running down the center of each petal. Similar to the native floatinghearts, the primary method of reproduction for crested floatingheart is via spiky ramets, which float through the water column and eventually sink to the bottom where they can root and sprout to form new plants. A single founder plant can produce as many as 500 ramets over six months and 40% of these are likely to sprout, so ramets have the potential to start completely new infestations in a relatively short amount of time. Seed production by crested floatingheart is not well-understood but this species has reportedly hybridized with bananalily and therefore seems able to reproduce via seeds.

Due to its attractive appearance, crested floatingheart has long been grown as a water garden ornamental. The species was first discovered in the wild in 1996, when it apparently escaped cultivation in southwest Florida, invaded the lake of a residential subdivision, and quickly spread throughout the drainage canals. By 2001, crested floatingheart had spread north to Sarasota and east to Palm Beach County and is now broadly distributed throughout the state, which spurred its addition to the Florida Noxious Weed List in 2014. The species was found in South Carolina's Santee Cooper system in 2006 and continues to be one of the most vexing problems for resource managers there. In addition to Florida and South Carolina, active invasions of crested floatingheart exist throughout the southeastern United States (eastern Texas, Louisiana, North Carolina and possibly Mississippi).

Yellow floatingheart (*N. peltata*) is native to Central Europe and Asia Minor, where it occurs in temperate and subtropical climates. Leaves are oval to heart-shaped with a wavy or scalloped margin and are attached to the stems in an opposite manner. Leaf size and shape are dependent upon season; plants produce small leaves in winter, and spring to early summer brings small folded leaves that open more when temperature and daylight increase. The upper surface of the leaves is green to yellow-green and the underside may be maroon or purple. A bright yellow five-petaled flower that measures up to 2 inches across is produced at each leaf. The flower has a broad membranous margin that is wavy to ruffled, which creates an irregular "fringe". Yellow floatingheart does not seem to produce ramets, but instead produces stolons that grow through the water column and along the bottom of slow-moving water bodies. Stolon fragments produce adventitious roots and form young plants with a single leaf, but the primary mode of reproduction for yellow floatingheart appears to be seed production. Each flower produces a seed capsule that splits at the end of the growing season and releases many smooth seeds with winged margins. The seeds float until a disturbance causes them to break the water surface; they then sink, remain dormant through the winter and sprout in the spring.

Yellow floatingheart is cosmopolitan in distribution and is found throughout much of the world. The species may be spread via by escaping cultivation or by hitchhiking on birds or other aquatic animals. It is invasive in the United States (where it has been reported in 34 states) and New Zealand, but threatened in Japan. Yellow floatingheart was first documented in 1882 in New York City's Central Park, where it was grown in a terrace pond, and has been marketed as an ornamental in the water garden industry since 1891. By 1930, it was found out of cultivation in the Pacific Northwest and was reported in eastern Washington and Oregon. Similar to crested floatingheart, yellow floatingheart was added to the Florida Noxious Weeds List in 2014.

Water snowflake (*N. indica*) is native to tropical Asia and Africa. The thick, fleshy, round leaves measure up to 7 inches across and are bright green on the upper and lower surfaces. Each flower has six to twelve white densely fringed petals that are covered with hairs, and multiple flowers may be associated with a single leaf. Water snowflake reproduces mainly via seed but can also propagate very effectively through vegetative means such as stem fragments.

Water snowflake was introduced either intentionally or as an accidental escape from the water garden and aquarium industry. Water snowflake has been vouchered in several counties in Florida, although most of those specimens have now been identified as Humboldt's floatingheart (see below) through molecular genetics. The only other report of non-cultivated water snowflake in the United States is from Hawaii, but the status of that infestation is unknown.

Floatinghearts of uncertain origin

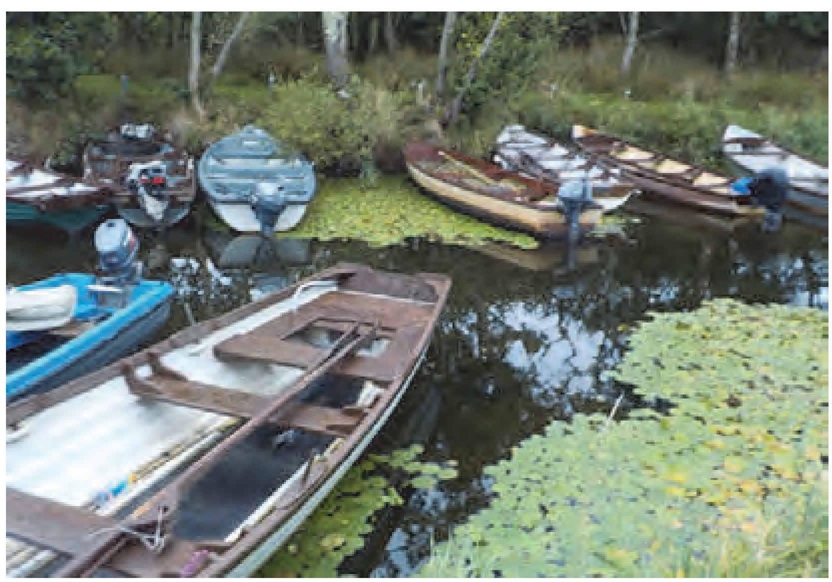
Gray's floatingheart (*N. grayana*), native to Cuba and the Bahamas, was recently identified in Florida. The USDA classifies the species as non-native and Wunderlin et al. (2019) states the plant is probably a water garden escape in Florida, where the species has been reported in three counties. It is likely that the species has been in Florida for a number of years but was misidentified as a color variant of water snowflake. Gray's floatingheart has arrowhead- or heart-shaped leaves that are dull green on the upper surface and purplish on the underside with overlapping lobes. The flowers have bright yellow petals with densely fringed margins. Information regarding the reproductive ecology and true native range of Gray's floatingheart is uncertain due to lack of information about this species.

Humboldt's floatingheart (*N. humboldtiana*) has also been recently described in Florida. The species is native to the Caribbean and Mexico and is classified as introduced (non-native). Humboldt's floatingheart has oval to heart-shaped leaves that are bright green (or sometimes mottled) on the upper surface and underside with indistinct veins. The white flowers are very similar to those of water snowflake but usually have five petals. Humboldt's floatingheart reproduces via seeds that lack tubercles (small, wart-like projections). This characteristic can be useful to separate this species from water snowflake, which produces seeds that do have tubercles. Humboldt's floatingheart has been reported in two counties in Florida and one county in Texas. As with Gray's floatingheart, it is likely that Humboldt's floatingheart has been in Florida for a number of years but was misidentified as a variant of water snowflake.

Reproduction

Reproduction occurs via ramets in some species or through the production of seeds in others. As mentioned earlier, ramets are rhizome-like clusters that form at the base of the leaf and eventually break off and have the potential to form new plants. Several species of *Nymphoides* are also able to root from stem or leaf fragments, while a few utilize a combination of these strategies. Ramets may float away from the parent, which can increase population size, or may settle to the bottom of the water body, where they remain until they sprout. Seeds of some species of floatinghearts may have a dormancy period, while others may readily germinate almost as soon as they are shed by the parent plant. Plant fragments, ramets and seeds may be spread via currents, animals or via human activities by hitchhiking on recreational equipment.

Problems associated with *Nymphoides*



Introduced species of *Nymphoides* such as crested and yellow floatinghearts have the potential to interfere with navigation, recreation, flood control and other water-based activities by clogging the aquatic system with dense plant material. These invasive floatinghearts species cause ecological problems as well. Their accelerated reproduction and ability to create canopy-forming monocultures on the water surface allows these species to outcompete and displace native floatinghearts and other desirable indigenous species, thus reducing habitat quality. In addition to these immediate ecological effects, it appears that several species of this genus are able to hybridize; for example, hybridization has occurred between two native species (little floatingheart and bananalily) and between native and invasive species (crested floatingheart and bananalily). These hybridization events could eventually cause significant contamination of the gene pool for native floatinghearts, which would imperil the integrity of these important plants.

Management options

It should be clear that the varied reproductive strategies (ramets, seeds and fragmentation) of introduced floatinghearts are problematic for resource managers and may limit the use of some management techniques. Similar to many introduced species, the best method to prevent these plants from causing ecological damage and interfering with human uses of an aquatic resource is to prevent the species from entering the water body. Some states do prohibit the sale and transport of some species of floatingheart, but existing populations still manage to slowly increase their range and colonize new bodies of water.

Physical (Section 3.4) or mechanical (Section 3.5) control measures such as hand removal or mechanical harvesters are not practical for managing crested and yellow floatinghearts because both species can reproduce via stem and leaf fragments and are thought to have the capacity to regrow from root crowns that remain in the soil after harvesting plant material in the water column. In addition, crested floatingheart produces many ramets and yellow floatingheart produces copious seeds, both of which are left behind after harvesting and can quickly sprout to repopulate the system. Cultural control methods (Section 3.4) such as dewatering or drawdowns may have some utility for crested floatingheart management, as ramet viability is significantly decreased after 24 hours of desiccation. However, the seeds of some floatingheart species are induced to germinate by drawdown, which can result in significant infestations once water levels are returned to their normal state. Ramets that are buried under as little as 4 cm of soil rarely sprout, so benthic barriers may reduce the number or density of populations that are derived from ramets. No floatingheart-specific biocontrol agents (Section 3.6) have been identified for these species, although the aquatic larvae of a native generalist moth reportedly consumes native floatinghearts.

The most effective tool in the toolbox for floatingheart management is chemical control (Section 3.7.1). Although both foliar and water-column treatments can be utilized to control large infestations, water-column treatments tend to have better efficacy. Contact herbicides such diquat and endothall and systemic herbicides such as imazamox, imazapyr, and auxins (2,4-D, triclopyr and florpyrauxifen-benzyl) have been used for management of floatinghearts with varying levels of success. As with other floating-leaved species, herbicide efficacy is influenced by environmental conditions such as currents and wave action.

Summary

The genus *Nymphoides* is quite diverse and the United States hosts both native and introduced species. Introduced species – especially crested and yellow floatinghearts – can have serious effects on the environment by outcompeting native floatinghearts and other desirable indigenous species and can interfere with human uses of invaded waters by clogging the water column and inhibiting flow. Early detection and rapid response is the least expensive and most effective means to prevent introduced floatinghearts from expanding into uninvaded aquatic systems. Aggressive management should be employed in areas where these introduced species already occur to avoid expansion of existing populations and reduce the likelihood of new introductions to connected waterways. The use of most control methods is limited due to the multiple reproductive strategies utilized by introduced floatinghearts, but several chemical control tools can be used to manage populations of invasive members of the genus *Nymphoides*.

Photo and illustration credits:

Page 59: Crested floatingheart in a canal in Naples, Florida; Lyn Gettys, University of Florida
Page 60 upper: Bananalily (inset: ramet); Lyn Gettys, University of Florida
Page 60 lower: Crested floatingheart (inset: ramet); Lyn Gettys, University of Florida
Page 61 upper: Yellow floatingheart (inset: seed capsules); Lyn Gettys, University of Florida
Page 61 lower: Water snowflake; Lyn Gettys, University of Florida
Page 62 upper: Gray’s floatingheart; Lyn Gettys, University of Florida
Page 62 lower: Yellow floatingheart infestation in Ireland; Lyn Gettys, University of Florida
Page 63 upper: Adventitious roots on yellow floatingheart stems; Lyn Gettys, University of Florida
Page 63 lower: Humboldt’s floatingheart mother plants and seedlings in a dry canal in Puerto Rico; Lyn Gettys, University of Florida

2.10 Waterchestnut

Scott A. Kishbaugh (retired) and Catherine McGlynn: New York State Department of Environmental Conservation, Albany NY; scott.kishbaugh@dec.ny.gov and catherine.mcglynn@dec.ny.gov

Trapa natans L; floating-leaved plant in the Trapaceae (waterchestnut) family; originally placed in the Hydrocharitaceae (frog's-bit) family, but at the molecular level should be placed in the Lythraceae (purple loosestrife) family
Derived from *calcitrapa* [Latin: a spiked iron ball ("caltrops") used as an ancient weapon] and *natans* (Latin: swimming)

Introduced from Asia to Massachusetts and New York in the late 1870s to early 1880s
Present in the mid-Atlantic into the Northeast, south to northern Virginia, west to central Pennsylvania, east to New Hampshire, north to Quebec

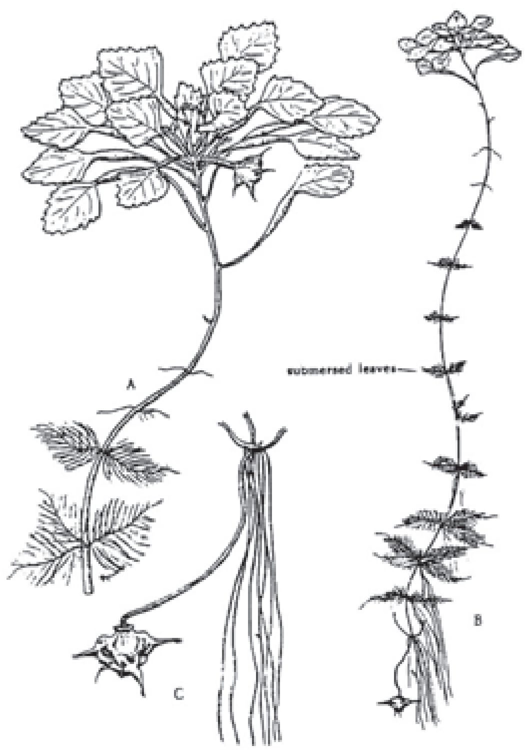
Introduction and spread

Botanists have subdivided the genus *Trapa* into at least 20 to 25 different species based upon small differences in the nutlets. Under the most recent taxonomic schemes, *Trapa natans* is subdivided into three varieties. The varieties *Trapa natans* var. *bispinosa* and *Trapa natans* var. *bicornis* are found primarily in northern India and southeastern Asia, where both are grown as agricultural crops, whereas the variety *Trapa natans* var. *natans*, commonly called waterchestnut, is a prized agricultural crop in India and China, a protected and disappearing plant in Europe and a highly aggressive

invader in the United States. Waterchestnut is often confused with the Chinese waterchestnut (*Eleocharis dulcis*), an edible tuber common in Chinese cuisine. Both species have been widely cultivated as a food source, but they are unrelated. Although "waterchestnut" is the most widely used common name for *Trapa natans* var. *natans*, the variety is also known by a number of other common names, with religious ("Jesuit's nut"), evocative ("water caltrops", "bull nut") and sinister ("devils nut", "death flower") connotations.

Waterchestnut is native to Eurasia and Africa and archaeologists have found evidence of waterchestnut in sediments dating back to at least 2800 BC. The first introduction of waterchestnut to the US is better documented than that of most other exotic plants, but there is some debate regarding the specific time and place of this introduction. The initial introduction to North America was well-described by Eric Kiviat in a Hudsonia newsletter. North American infestations can probably be traced to two distinct locations. Waterchestnut was first introduced from Europe to Middlesex County, Massachusetts around 1874 and was cultivated as an ornamental in Asa Gray's botanical garden at Harvard University in 1877. Seeds were distributed by Harvard gardeners into nearby ponds over the next several years; as a result, waterchestnut migrated into the Concord and Sudbury Rivers by the mid-1880s, reached nuisance portions by the turn of the century and underwent explosive growth by the 1940s.

Another introduction occurred in Scotia in eastern New York during the early 1880s. A Catholic priest planted waterchestnut seeds from Europe in Sanders Pond (now Collins Lake), which led to extensive colonization of the lake by 1884. Subsequent flooding of the neighboring Mohawk River (via locks and dams on the New York Barge Canal) further spread the plant and spawned widespread growth by the 1920s. Waterchestnut was reported in the Hudson River by 1930 and reached nuisance levels in the 1950s. The species likely then spread west through the Erie Barge Canal system and reached Oneida Lake and the Finger Lakes region by the turn of the 21st century. Waterchestnut also migrated north into Lake Champlain through the Hudson-Champlain Canal and most likely reached Quebec through the Richelieu River system during the late 1990s. Waterchestnut was first found in Maryland in the late 1910s and reached the Potomac River during the early 1920s; widespread populations were present by the 1940s. As of 2018, the waterchestnut range still appears to be limited to the northeastern United States, although populations may have been extirpated in Delaware and the District of Columbia.



Description of the species

Waterchestnut is an ideal candidate for early detection programs because its appearance differs from all other plants found in North America and the species can often be identified early in its colonization cycle. Waterchestnut is an annual floating-leaved dicot that grows primarily in sluggish, shallow water. The habitat for this species includes lakes, ponds, reservoirs, sheltered margins of flowing water, freshwater wetlands and fresh to brackish estuaries. Waterchestnut usually grows in water less than 7 feet deep but has been found at depths of 12 to 15 feet. The species prefers thick, nutrient-rich organic sediments and an alkaline environment, but is tolerant of a wide pH range. Waterchestnut will not grow in salt water, although it can survive in brackish water with freshwater springs and groundwater input. The species grows aggressively and regularly produces as much as one pound of dry weight per square yard of surface area. Severe infestations can result in much greater biomass production; for example, waterchestnut populations growing in shallow impoundments in upstate New York have reportedly yielded almost 17,000 pounds of dry biomass per acre.

Submersed leaves of waterchestnut are pinnate (feather-like) and superficially resemble the finely dissected leaves of milfoils (*Myriophyllum* spp.). Submersed leaves are up to 4inches long and are attached to the flexible stem in a whorl. Surface or floating leaves are palmate (divided like the fingers on a hand) and form a rosette that can be as broad as 1 foot in diameter. Leaf blades are 1 to 2 inches long and diamond shaped with a coarsely serrated (saw-toothed) margin. The upper sides of the leaves are bright green and the undersides are yellow-green with prominent veins. Rosettes form below the water surface and elongate to the surface by late spring – plants are buoyant due to inflated petioles or leafstalks (bladders) just below the rosette of leaves. Surface rosettes can form and may initially be hidden within beds of other plants that produce floating leaves [e.g., watershield (*Brasenia* spp.), spatterdock (*Nuphar* spp.) and white waterlilies (*Nymphaea* spp.)] and by smaller floating plants such as duckweed, watermeal (Section 2.14) and filamentous algae (Section 2.18). However, the prolific growth of waterchestnut will eventually create dense monocultures with as many as 50 rosettes per square yard and will crowd out desirable native plants. Waterchestnut beds can be so extensive that they may form up to three layers that completely cover the shallow zones of lakes and rivers and may obscure the margin between land and water.

Waterchestnut produces a single-seeded four-pronged nutlet with barbed spines. This structure is only produced by *Trapa natans* var. *natans* and allows for easy identification of the variety. The barbed spines are sharp enough to penetrate a wet suit – a painful experience for anyone unfortunate enough to step on one of these nutlets – and are the basis for the imaginative common names given to this plant. In addition to wreaking havoc on divers and swimmers, these nutlets figure prominently in the spread and propagation of this invasive species.

Reproduction

Many invasive species spread and reproduce from fragments, tubers, turions or underwater runners or stolons, but waterchestnut is an annual that reproduces solely from seeds. Small white flowers with yellow stamens are produced on the rosette after June, then drop into the water during summer and mature as nutlets between July and September. Each rosette produces 10 to 20 nutlets, which are capable of persisting for 10 to 15 years if kept moist in nutrient-rich sediment. Nutlets are around 1 inch wide, approximately 20% more dense than water and change from fleshy green to woody black by late summer. Mature nutlets drop from the plant and quickly sink into the sediment or wash to the shoreline, where the barbed spikes anchor the nutlet into the sediment. Parent plants disintegrate in the fall and seeds begin to germinate within a month after water temperatures warm to 50 °F or higher the following spring. A single nutlet can produce multiple rosettes (up to 15 to 20 surface rosettes) because the rhizome can branch laterally to produce multiple upright stems.

Nutlets migrate between bodies of water by a variety of means. The most conspicuous vector for many years was humans, who intentionally introduced the waterchestnut as an ornamental. *Trapa natans* is listed as a federal “species of concern”, but there are currently no explicit federal transport restrictions. Fortunately, a new appreciation of the environmental and economic problems that accompany establishment of this species and a network of state laws (including laws in New York, Vermont, New Hampshire, Illinois, Michigan, Florida, Minnesota, Wisconsin and Maine) that prohibit its transport have greatly reduced intentional introduction of waterchestnut. However, nutlets continue to move on currents between connected waterways, on the feathers, talons and webbed feet of numerous waterfowl and furred mammals, and especially on boat propellers, trailers and even foam bumpers on canoes.

Problems associated with waterchestnut

Infestations of waterchestnut cause problems similar to those of other invasive aquatic plants. Waterchestnut provides little wildlife value and can form dense surface canopies that reduce sunlight penetration into the water column by 95% and crowd out other submersed and floating-leaved native plants and the fauna that rely on these plants for food and shelter. There is strong evidence that water celery (*Vallisneria americana*), a highly valued native plant, has been eliminated from many parts of the Hudson River after colonization by waterchestnut. This is due to the reduction in habitat available to water celery and to depletion of dissolved oxygen under large waterchestnut canopies, which also has a negative effect on small invertebrates. Large populations of waterchestnut create hostile environments for many desirable species such as banded killifish and spottail shiner and are often inhabited by fauna that are more tolerant of adverse conditions, including rough fish species such as the common carp. There is some evidence that waterchestnut inhibits the growth of phytoplankton due to production of allelopathic substances. Dense beds of waterchestnut can also entrap predatory birds seeking food within and underneath the surface canopy. Although waterchestnut canopies could potentially create significant pockets of still water to support mosquitoes, this has not been well documented in North American populations of waterchestnut.

Waterchestnut often grows under eutrophic conditions (Section 1.1), in part because eutrophic bodies of water often create the thick organic sediments preferred by this plant and in part because waterchestnut grows in shallow waters where poor water clarity does not limit plant growth. Thick masses of leaves and stems generated by waterchestnut degrade and settle into the bottom sediments, which increases the organic content (and depth) of the sediment and contributes to greater turbidity and a cycle of increasing eutrophication. Bacterial degradation of this plant material can reduce dissolved oxygen, particularly at the end of the daily respiration cycle and when plants rapidly degrade in response to active management, such as herbicide treatment. Plant tissues also accumulate some heavy metals; this may occur with other highly abundant aquatic plants as well and may ultimately be a net benefit since these metals are removed from sediments or the water column.

Dense surface canopies of waterchestnut reduce water flow and impede boating and other forms of non-contact recreation, a particularly vexing problem since this plant often dominates navigable rivers and slow-moving water around marinas. Unlike submersed invasive plants and most floating-leaved plants, waterchestnut creates canopies that are impenetrable by even canoes and kayaks – the rosettes swallow paddles and significantly retard the momentum of the paddler. The same shallow waters frequented by canoers and kayakers are sometimes used for swimming, although the soft, thick organic sediments usually needed to support waterchestnut plants do not provide the ideal habitat for waders and swimmers. Waders willing to slog through dense populations of waterchestnut must carefully navigate through the nutlets commonly found along the shoreline and in the upper layer of near-shore sediments since stepping on the sharp barbs can cause deep puncture wounds. Dense mats create an additional safety concern – entanglement in waterchestnut beds may have contributed to drowning deaths in the Hudson River in 2001.

The most significant impact of waterchestnut infestations on humans may well be a reduction in aesthetics. Dense waterchestnut beds can completely cover the surface of shallow bodies of water and small ponds and will often carpet the near-shore areas of popular navigable rivers. The description grudgingly applied to waterhyacinth (Section 2.11) – “chokes out a water surface” – applies to waterchestnut as well.

Management options

During the past 100 years, many techniques have been used to manage waterchestnut. Unlike most invasive aquatic plants, waterchestnut has been effectively controlled and perhaps even eradicated in some bodies of water, but only after persistent effort. Similar to other invasive plants, best management of waterchestnut results from a vigilant prevention program. Weed watcher programs are particularly effective in controlling waterchestnut since the species is easily identified and early intervention greatly improve opportunities for eradication.

Once present in a body of water, waterchestnut can be controlled by physical and chemical techniques and may ultimately be managed by biological agents. Initial infestations, particularly when only a single rosette is found, can be pulled by hand (Section 3.4). The best window for removal of waterchestnut is from mid-June to mid-August – earlier efforts may result in regrowth or incomplete removal of nutlets, whereas later attempts might miss some nutlets or cause loosely attached seeds to dislodge. Plants should be flipped upside down immediately after removal to prevent dropping of seeds. Kayaks or canoes can be used for hand removal of waterchestnut; kayaks are more easily maneuvered through dense beds of waterchestnuts, but canoes carry more chestnut cargo. Hand removal programs led by cooperative extension offices, community groups, Boy Scout troops and volunteers have effectively controlled waterchestnut in Oneida Lake in central NY and in many other smaller bodies of water throughout the Northeast.

Mechanical harvesting (Section 3.5) can effectively control large infestations of waterchestnut since the species does not reproduce by fragmentation, although cutting just the leaves (rosettes) from plants will likely leave nutlets in the system. Mechanical harvesting of plants after seeds have formed but before they mature can effectively break the reproductive cycle of the plant; however, the longevity and quantity of seeds in the sediment’s seed bank may make it

necessary to repeat the operation for at least 5 to 10 years to eradicate the species. A variety of state and federal agencies have used large mechanical harvesters to greatly reduce waterchestnut populations in Lake Champlain in Vermont and New York and in the Mohawk and Potomac Rivers. However, populations rapidly rebounded and returned to pre-harvesting densities if harvesting is suspended before a population is extirpated.

Herbicides have also been used to control large-scale infestations of waterchestnut (Section 3.7.1). The herbicide used most often for control of this aquatic weed is 2,4–D, which is usually applied in early summer when plants are just reaching the water surface. Recently, triclopyr has also been used to control waterchestnut. Research is underway to determine whether glyphosate provides control of waterchestnut when applied directly to the rosette of surface leaves. Some formulations of imazamox has also been used to control waterchestnut.

Grass carp (Section 3.6.2) have been used as biocontrol agents to manage waterchestnut in some bodies of water. However, grass carp are relatively indiscriminate feeders that find waterchestnut to be unpalatable, so few plants are consumed. Insect-based biocontrol (Section 3.6.1) may be a more promising alternative; researchers continue evaluating a native leaf beetle (*Galerucella birmanica*) which has shown promise. However, this native beetle is a generalist feeder that consumes plants other than waterchestnut. Because successful biocontrol agents must be species-specific and feed only on a particular host plant (Section 3.6), this native beetle may not be a viable biocontrol option for waterchestnut, although some biocontrol work on waterchestnut has been funded by New York state. Some research has also evaluated the use of ultrasonic waves at 20 kHz to break down plant tissues, but this work has not translated to large scale control efforts.

Summary

Waterchestnut is one of the most invasive aquatic plants in the northeastern United States and has spread from its introduced range into neighboring states over the last 125 to 150 years. This species creates significant ecological damage, restricts human use of waterways and can be very difficult to control without consistent and persistent effort. However, waterchestnut is unique among invasive aquatic plants because it is easily detectable through citizen watch programs and can be controlled or even eradicated if caught early in its colonization. The species is an annual and can be managed by preventing seed production. Once established, waterchestnut requires significant resources to manage and vigilant use of mechanical or chemical control methods for 10 to 15 years to exhaust the reservoir of dormant seeds harbored in sediments.

Photo and illustration credits:

Page 65: Volunteers with the Buffalo Niagara Riverkeepers removing waterchestnut from Ellicott Creek Park in Western New York; Mike Goehle, USFWS
Page 66: Line drawing; Barre Hellquist. From Crow GE and CB Hellquist. 1983. Aquatic vascular plants of New England: part 6. *Trapaceae, Haloragaceae, Hippuridaceae*. Station Bulletin 524. New Hampshire Agricultural Experiment Station, University of New Hampshire, Durham, NH
Page 67: Waterchestnut nutlets; TheDarkCurrent, distributed under a CC BY-SA 3.0 license.
Page 68: Waterchestnut plant; Hilary Smith, The Nature Conservancy
Page 69: Mechanical harvesting waterchestnut from Tonawanda Creek in Western New York; Mike Goehle, USFWS

2.11 Waterhyacinth

Lyn A. Gettys: University of Florida FLREC, Davie FL; lgettys@ufl.edu

Pontederia (*Eichhornia*) *crassipes* (Mart.) Solms; floating plant in the Pontederiaceae (pickerelweed) family
Derived from *Eichhorn* [Johann Albrecht Friedrich Eichhorn (1779-1856), Prussian minister of education and public welfare] and *crass* (Latin: thick)
"plant with thick leaf stalks"

Introduced from Brazil to New Orleans in 1884
Present throughout the southeastern US and California, Hawaii and the Caribbean area

Introduction and spread

Pontederia crassipes (formerly *Eichhornia crassipes*) is native to the Amazon River and has been widely introduced throughout the tropical regions of the world, most recently occurring in Lake Victoria in East Africa. The first known introduction of waterhyacinth to North America was at the Cotton States Exposition in New Orleans in 1884. The species was initially cultivated as an ornamental but quickly escaped cultivation and invaded other parts of the

southeastern US. Waterhyacinth must have been a botanical curiosity due to its size, floating growth habit and the beauty of its very short-lived purple flower spikes. Mr. Fuller (the owner of Edgewater Grove, 7 miles upstream of Palatka on the St. Johns River) introduced this “beauty” to Florida around 1890. It was initially grown in Mr. Fuller’s fountain pond and excess growth was cast into the St. Johns River, where within a short time it covered the half-mile wide river from bank to bank at several locations. Waterhyacinth spreads very rapidly; for example, the species covered 126,000 acres of Florida’s surface water within 70 years of its arrival in that state. Waterhyacinth is present throughout the southeastern US, California, Hawaii and the Virgin Islands, but is considered eradicated in Arizona, Arkansas and Washington State. Populations of waterhyacinth have been reported in other states, including New York, Minnesota, Kentucky, Tennessee and Missouri and plants are intentionally introduced to farm fish ponds in southern Arizona and southern Delaware. This species is not cold-hardy and has not established permanent populations in more temperate areas outside the southern US. Waterhyacinth will survive moderate freezes but requires temperatures of greater than 50 °F to produce new growth. A number of states, including Florida, South Carolina and the US territory Puerto Rico, prohibit the sale of waterhyacinth, but the species is still available for purchase from aquarium supply stores, aquatic plant nurseries and internet sources in other states. Waterhyacinth spreads in natural systems by producing seedlings and daughter rosettes – small plantlets that are attached to the mother plant by a floating stolon or runner. Rosettes can easily become caught in boat trailers or live wells, which results in the introduction of the species to new bodies of water. Waterhyacinth is also spread by uninformed water garden and pond owners, who (along with Mr. Fuller in the 1890s) believe they are beautifying canals and lakes by tossing extra plants into natural systems.

Description of the species

Waterhyacinth is a floating flowering monocot that grows as an annual (in temperate regions) or as a perennial (in tropical and subtropical climates) in all types of bodies of water. Muddy or turbid water often limits growth of submersed plants, but because waterhyacinth is a floating plant, it is unaffected by these conditions. The leaves of waterhyacinth are thick, glossy, waterproof and rounded with a heart-shaped base. Each leaf can reach up to three feet in length and is borne singly on a spongy, inflated petiole (leaf stalk). Leaves are attached to one another at the base of the petiole to form a rosette that is free-floating, although plants will sometimes root in soft saturated sediments when stranded by drought or wave action. The dark purple to black roots of waterhyacinth are long and feathery and hang beneath the rosette of leaves. Waterhyacinth grows throughout the year in the tropics, but freezing temperatures kill the leaves of the plant in the northern portions of its range. Cold-damaged leaves then fold down and protect the meristem, which grows at or immediately below the surface of the water.

The most striking feature of waterhyacinth is the spike of large, showy flowers produced from the center of the rosette of leaves. Flowers are borne in groups of 8 to 15 on a single spike that can rise up to 20 inches above the rosette. Each flower is up to 3 inches tall and has six lavender-blue to purple petals, with the uppermost petal marked by a yellow "eye-spot". Flowers are short-lived, with each lasting only one or two days, but a spike may be showy for up to a week since only a few flowers open each day. Flowering is indeterminate – flowers at the base of the spike open first and flowers at the top of the spike open last. After flowers are fertilized, the spike bends and dips into the water, where many tiny seeds are produced in capsules. Mature seeds drop to the bottom of the body of water, where they remain dormant until sediments are exposed after water levels fall due to drought.

Waterhyacinth is sometimes confused with native frog's-bit (*Limnobium spongia*), because both are floating plants with rounded leaves borne in rosettes. However, the roots of waterhyacinth are black and feathery, whereas the roots of frog's-bit are thicker and white. In addition, the petioles of frog's-bit are usually slender, while the petioles of waterhyacinth are often spongy and bladder-like. Finally, flowers of frog's-bit are small, white and much less showy than those of waterhyacinth.

Reproduction

Waterhyacinth spreads by both seed and vegetative reproduction. As noted above, seeds are tiny and remain dormant until conditions are favorable for germination. Some reports suggest that seeds germinate best after they have dried and others say that seeds must be exposed to alternating warm and cold temperatures before they will germinate. Seed reproduction can be important in temperate climates since waterhyacinth is killed by freezing temperatures and recolonization in spring may be dependent on the seed bank established during the previous growing seasons. Once seeds have germinated and conditions are favorable for growth, waterhyacinth rapidly produces new daughter plants, or ramets, from horizontally growing stolons. Daughter plants can be produced in as little as 5 days under optimal growing conditions and populations can double in size in as little as 6 to 18 days, so the rapid growth and spread of waterhyacinth is due primarily to this type of vegetative reproduction.

Problems associated with waterhyacinth

Waterhyacinth grows almost entirely on the surface of the water as a floating plant and its growth potential is limited only by temperature and the availability of nutrients. Waterhyacinth prefers an environment similar to that favored by desirable fish populations – mesotrophic and eutrophic habitats with an adequate supply of calcium and a pH ranging from 6.5 to 9.5. There is no doubt that waterhyacinth is a serious aquatic weed. Under optimum conditions, an undisturbed population of waterhyacinth is composed of about 10 plants per square foot and has a fresh weight of 10 pounds. An acre (43,560 square feet) of waterhyacinth would therefore be home to about 435,600 plants with a fresh weight of around 200 tons. Since 95% of the plant weight is attributable to water, only 5% of the fresh weight – about 10 tons per acre – remains after plants are harvested and dried.

Waterhyacinth may not be as productive as most agricultural crops; however, trying to remove or stop 200 tons of live waterhyacinths from jamming against a bridge or clogging a waterway is no simple task! Large colonies of linked mother and daughter plants form dense rafts or mats that can quickly cover a body of water from shore to shore. Left undisturbed, floating mats of waterhyacinth provide a perfect substrate or "island" to support the growth of additional grasses, herbaceous plants and even small trees, which further bind the floating mat together. These mats interfere with human use of waters. For example, large populations of waterhyacinth can restrict recreational and commercial activities and can make boating, fishing and swimming impossible. In addition, water flow is greatly reduced where mats of waterhyacinth are present, which can impede irrigation and flood control efforts. Infestations of waterhyacinth can have serious ecological impacts as well. Dense waterhyacinth populations also reduce species richness or plant diversity by limiting light availability to native submersed plants and by crushing communities of emergent plants along the shoreline. The loss of these plants also eliminates habitats for animals that depend on native plants for shelter, nesting and food. In addition, large mats block the air-water interface and reduce dissolved oxygen, which makes the system uninhabitable to fish and other aquatic fauna.

Management options

The best method to control waterhyacinth is to prevent the species from entering a water body. The sale and interstate shipment of a closely related species [rooted waterhyacinth (*E. azurea*)] is prohibited by the Federal Noxious Weed List and its introduction into the US has been avoided thus far. Waterhyacinth (*P. crassipes*) is not on the Federal Noxious Weed List because the species was already widely distributed in the US at the time the Federal Noxious Weed Acts were developed. In spite of these prohibitions, waterhyacinth still manages to slowly increase its range and to colonize new bodies of water.

Physical (Section 3.4) or mechanical (Section 3.5) control measures such as hand removal or mechanical harvesters should be designed to prevent the spread of waterhyacinth plantlets to other parts of the water body. Hand removal is labor-intensive and typically involves raking plants to the shoreline or into a boat. This very laborious task can seem deceptively easy; a pond that is a single acre in size may look small, but can host up to 200 tons of waterhyacinth that must be pulled out by hand! Plants are then offloaded along the shoreline until they desiccate and die. Hand removal may be an effective means to control waterhyacinth in small ponds, but is not practical in larger systems. Mechanical harvesting is usually used to remove plants from larger systems and involves heavy machinery that ranges from a backhoe on a barge to specialized equipment. A problem associated with mechanical harvesting of waterhyacinth is disposal of the harvested plants. Waterhyacinth vegetation has been used to make furniture, baskets and other items in some parts of the world and has been evaluated for its potential as mulch, cattle feed, biofuel production and other uses, but its utility is very limited. As a result, most harvested waterhyacinth is generally disposed of in farm fields or a landfill. Hand removal of waterhyacinth from ponds is best employed after herbicide application has been used to control the majority of the plants. Regular removal of missed plants and any plants growing from seeds after herbicide treatment will prevent waterhyacinth from reinfesting the pond.

Drawdowns can be used to “strand” and desiccate waterhyacinth on exposed shorelines, but the time required to effectively dry large mats of plants can be long. Also, drawdowns and drought have been known to trigger seed germination and plants reestablish quickly when water levels rise. Therefore, most waterhyacinth management programs in the US rely on the use of herbicides in conjunction with established insect biocontrol agents. Waterhyacinth weevils (*Neochetina* spp.) were introduced and established in the early 1970s (Section 3.6.1). The weevils are found throughout the range of waterhyacinth but in most areas the insects only slow plant growth and reproduction and do not provide adequate control of the weed. As a result, herbicides are used in maintenance control programs to keep plant populations low and to reduce growth potential of waterhyacinth. Herbicide selection is based on water use, selectivity to reduce damage to nontarget native plants and cost (Section 3.7.1). Several herbicides are commonly used as foliar sprays to selectively control waterhyacinth. Contact herbicides – including diquat, flumioxazin and endothall – are quickly absorbed by plant tissue and are fast-acting, whereas systemic herbicides – including 2,4–D, glyphosate, imazamox, penoxsulam and bispyribac – provide slower but effective control.

Summary

Waterhyacinth is one of the world’s worst aquatic weeds and causes problems in all tropical and subtropical continents. Its current distribution in the US is primarily from East Central Texas to the Atlantic Coast and north to coastal North Carolina. It also occurs in the Sacramento River Delta in California. Although waterhyacinth is occasionally found north of the central US, the species typically does not persist where waterways are subject to ice formation and prolonged freezing temperatures. Florida and the Gulf states are particularly impacted by waterhyacinth due to the moderate climate and shallow, naturally nutrient-rich lakes, but the species can colonize virtually any region in North America where winter temperatures remain above freezing and mesotrophic or eutrophic waters are present. Aggressive maintenance control programs have kept populations of waterhyacinth in check in most areas, but these efforts must be employed on a continual basis to avoid population explosions of this noxious invasive species.

Photo and illustration credits:

Page 71: Waterhyacinth infestation; University of Florida Center for Aquatic and Invasive Plants (photographer unknown)
Page 72: Waterhyacinth’s inflated petioles; Lyn Gettys, University of Florida
Page 73: Waterhyacinth clogging a south Florida canal; Lyn Gettys, University of Florida
Page 74: Flowers of waterhyacinth; Lyn Gettys, University of Florida

2.12 Waterlettuce

Lyn A. Gettys: University of Florida FLREC, Davie FL; lgettys@ufl.edu

Pistia stratiotes L.; floating plant in the Araceae (Arum) family
Derived from the Greek *pistos* (water) and *stratiotes* (a common soldier)

Introduction history uncertain, considered native to the southeastern US by some sources
Present throughout the southeastern US north to New Jersey and New York, west to Texas, Arizona and California; also present in Idaho, Ontario, Hawaii, Puerto Rico and the Caribbean

Introduction and spread

Pistia stratiotes is the only species in the genus *Pistia*. The origin of waterlettuce is unclear, but various sources suggest the plant is native to South America, Africa or the southeastern US. Waterlettuce is cosmopolitan in distribution and has been documented in aquatic systems around the world. The species is considered one of the world's worst weeds

and is a noxious species in most regions where it has been introduced, such as Hawaii, Australia and the Canary Islands. In addition, waterlettuce is considered invasive in the US, Puerto Rico and Africa, despite reports that the species could be native to these areas.

Fossil records show that waterlettuce was present in Africa, the species' center of diversity, 85 million years ago and that the plant was present in Florida at least 50 million years ago. The first modern report of waterlettuce in North America was made by John and William Bartram, who described dense, nearly impenetrable populations of the species while surveying the St. Johns River in Florida on New Year's Eve in 1765. The USDA considers waterlettuce to be native to the continental US and does not categorize the species as a noxious weed, but a number of state lists include waterlettuce as a noxious, invasive or prohibited plant.

Although not as productive as waterhyacinth (Section 2.11), waterlettuce spreads very rapidly and can double its population size in as little as a few weeks, so it can quickly cover the surface of invaded waters. The species is not cold-hardy and rarely establishes permanent populations in temperate areas. Waterlettuce will survive moderate freezes but requires temperatures of greater than 50 °F to produce new growth. A number of states – including Alabama, California, Connecticut, Florida, South Carolina and Texas – prohibit the sale of waterlettuce, but the species is still available for purchase from aquarium supply stores, aquatic plant nurseries and internet sources in other states. The species continues to inhabit many bodies of water in Florida, along with aquatic systems throughout most of the southeastern and southwestern US, Hawaii, Puerto Rico and the Virgin Islands. Despite the well-documented problems associated with waterlettuce, the species is still widely cultivated as an ornamental in water gardens and has been evaluated for its utility as a phytoremediation agent to reduce nutrients and heavy metals in contaminated waters.

Waterlettuce spreads in natural systems by producing seedlings and daughter rosettes – small plantlets that are attached to the mother plant by a floating stolon or runner. Rosettes can easily become caught on boat trailers or in live wells, which results in the introduction of the species to new bodies of water. Waterlettuce is also spread accidentally as a result of escapes from cultivation and intentionally by uninformed water garden and pond owners, who believe they are beautifying canals and lakes by tossing extra plants into natural systems.

Description of the species

Waterlettuce is a floating flowering monocot that grows as an annual (in temperate regions) or as a perennial (in tropical and subtropical climates) in all types of bodies of water. Muddy or turbid water often limits growth of submersed plants, but since waterlettuce is a floating plant, it is unaffected by these conditions. The leaves of waterlettuce have wavy or scalloped margins and are thick, light green, covered with short hairs and water-repellant. Each leaf can reach up to one foot in length; leaves are attached to one another at the plant's base to form a free-floating rosette (although plants will sometimes root in soft saturated sediments when stranded by drought or wave action). The white to tan roots of waterlettuce are long and feathery and hang beneath the rosette of leaves. Waterlettuce grows throughout the year in the tropics, but freezing temperatures kill the leaves of the plant in the northern portions of its range.

The flowers of waterlettuce are borne in a spathe and spadix arrangement. The greenish spadix, a spike-like structure in the center of the inflorescence that houses separate female and male flowers, is sheathed by the white spathe, a hairy leaf-like bract. Although other members of the Araceae family – including caladiums, peace lilies and anthuriums – are ornamental species that are prized for their showy inflorescences, the spathe and spadix of waterlettuce is small and inconspicuous. It was long thought that waterlettuce did not produce seeds and that all reproduction by the species was vegetative via the formation of daughter plants; however, it is now known that waterlettuce produces copious, viable seeds and that this strategy allows the plant to maintain a presence in areas where droughts or winter freezes kill mature plants.

Reproduction

Waterlettuce spreads by both seed and vegetative reproduction. Each plant produces multiple fruits and each 2 mm-long fruit can contain up to 20 tiny, golden-brown seeds. As a result, hundreds of seeds may be produced per square foot of coverage. Most seeds remain in the upper 2 inches of sediments and germination can be greater than 90% but it is not known how long these tiny seeds remain viable. Seed reproduction can be important in temperate climates since waterlettuce is killed by freezing temperatures and recolonization in spring may be dependent on the seed bank established during the previous growing seasons. Once seeds have germinated and conditions are favorable for growth, waterlettuce rapidly produces new daughter plants from horizontally growing stolons. In fact, the rapid growth and spread of waterlettuce during the growing season is due primarily to vegetative reproduction.

Problems associated with waterlettuce

Waterlettuce grows almost entirely on the surface of the water as a floating plant and its growth potential is limited only by temperature and the availability of nutrients. Waterlettuce prefers a habitat similar to that favored by desirable fish populations – mesotrophic and eutrophic waters with sufficient calcium and a pH ranging from 6.5 to 7.2. There is no doubt that waterlettuce is a serious aquatic weed, regardless of whether the species is native or introduced to the southeastern US. Under optimum conditions, a population of waterlettuce is composed of as many as 100 plants per square foot with a combined fresh weight of up to 5 pounds. An acre (43,560 square feet) of waterlettuce could therefore have millions of plants and a fresh weight of around 100 tons. Since 95% of the plant weight is attributable to water, only 5% of the fresh weight – about 5 tons per acre – remains after plants are harvested and dried.

Large colonies of linked mother and daughter waterlettuce plants form dense mats that can quickly cover a body of water from shore to shore and interfere with human use of waters. For example, large populations of waterlettuce can drastically impede boating, fishing and swimming and commercial activities. Also, water flow is greatly reduced where mats of waterlettuce occur, which hinders irrigation and flood control efforts. Several species of mosquito are known to breed in water held in the rosettes of waterlettuce; in fact, the larvae of some of these disease-causing insects attach to the underwater roots of waterlettuce and obtain oxygen through air tubes they insert into the plant's roots (Section 1.5). Infestations of waterlettuce can have serious ecological impacts as well. Dense waterlettuce populations reduce species richness or plant diversity by limiting the light that reaches native submersed plants and by crushing communities of emergent plants along the shoreline. The loss of these native plants also eliminates habitats for animals that depend on native plants for shelter, nesting and food. In addition, large mats block the air-water interface and reduce dissolved oxygen, which often makes the system uninhabitable to fish and other aquatic fauna.

Management options

The best method to control waterlettuce is to prevent the species from entering a water body. Waterlettuce is not on the Federal Noxious Weed List. However, waterlettuce is on the State Noxious Weed Lists of Alabama, California, Connecticut, Florida, Puerto Rico, South Carolina and Texas, so its sale and transport is prohibited in these states. Even in states where waterlettuce is listed, it is easy to purchase plants at farmers' markets, local plant sales, on the internet and from other unregulated sources. Although waterlettuce has been deemed eradicated in some invaded areas such as small field sites in New Zealand, it is difficult or impossible to completely eliminate waterlettuce once a larger body of water has been invaded. Between existing populations that are left uncontrolled, accidental transfer from infested areas and escapes from cultivation, waterlettuce still manages to slowly increase its range and to colonize new bodies of water.

Physical (Section 3.4) or mechanical (Section 3.5) control measures such as hand removal or mechanical harvesters should be designed to prevent the spread of waterlettuce plantlets to other parts of the water body. Hand removal is labor-intensive and typically involves raking plants to the shoreline or into a boat. This may seem like a simple job, especially in a small pond; however, a single acre can support as much as 100 tons of waterlettuce that must be pulled out by hand! Plants are then offloaded along the shoreline until they desiccate and die. Hand removal may be an effective means to control waterlettuce in small ponds, but is not practical in larger systems. Mechanical harvesting is usually used to remove plants from larger systems and involves heavy machinery that ranges from a backhoe on a barge to specialized equipment. A problem associated with mechanical harvesting of waterlettuce is disposal of the harvested plants. There are no large-scale uses of harvested waterlettuce, so most plant material is usually disposed of in farm fields or a landfill.

Drawdowns can be used to "strand" and desiccate waterlettuce on exposed shorelines, but the time needed to effectively dry large mats of plants can be long. Also, drawdowns and drought have been known to trigger seed germination of other invasive species such as waterhyacinth. Although there are as many as 50 species of insects that feed on waterlettuce, only two have met the criteria for biocontrol agents (Section 3.6.1). The waterlettuce leaf moth (*Spodoptera pectinicornis*) was imported from Thailand and released in Florida in 1990, but failed to establish. The waterlettuce leaf weevil (*Neohydronomus affinis*) was imported from South America to the US in mid-1980s and is now established throughout Florida, but its effect on waterlettuce growth is negligible. Therefore, most waterlettuce management programs in the US rely on the use of herbicides to keep plant populations low and to reduce growth potential of waterlettuce. Herbicide selection is based on water use, selectivity to reduce damage to non-target native plants and cost. Several herbicides can be used as foliar sprays to selectively control waterlettuce (Section 3.7.1). Contact herbicides such as diquat, carfentrazone and flumioxazin are quickly absorbed by plant tissue and cause obvious damage within a few days, whereas systemic herbicides such as imazapyr, penoxsulam and bispyribac provide slower but very effective control. Submersed application of the contact herbicide flumioxazin is currently being evaluated for selective control of waterlettuce, as are topramezone and the ALS herbicides.

Summary

Waterlettuce is one of the world's worst aquatic weeds and causes problems in virtually all waters it has invaded. It is currently distributed throughout the southeastern US north to New Jersey and New York, west to Texas, Arizona and California. While waterlettuce is found throughout New England and other temperate regions, it typically does not persist where waterways are subject to ice formation and prolonged freezing temperatures. Florida and the Gulf states are particularly impacted by waterlettuce due to the moderate climate and shallow, naturally nutrient-rich lakes, but the species can colonize virtually any region in North America where winter temperatures remain above freezing and mesotrophic or eutrophic waters are present. Aggressive maintenance control programs have kept populations of waterlettuce in check in most areas, but these efforts have to be employed on a continual basis to avoid population explosions of this noxious invasive species.

Photo and illustration credits:

Page 75: Infestation of waterlettuce; Lyn Gettys, University of Florida
Page 76: Long feathery roots of waterlettuce; Lyn Gettys, University of Florida
Page 77 upper: Spathe and spadix inflorescence of waterlettuce; Lyn Gettys, University of Florida
Page 77 lower: Young waterlettuce with daughter plant; Lyn Gettys, University of Florida

2.13 Giant and Common Salvinia

Linda S. Nelson: US Army Engineer Research & Development Center, Vicksburg MS (retired); Linda.S.Nelson@usace.army.mil

Salvinia molesta D.S. Mitchell; *Salvinia minima* Baker; free-floating ferns in the Salviniaceae family. Derived from *Salvinia* (after Antonio M. Salvini) and *molesta* (Latin: nuisance, annoying, troublesome) and *minima* (Latin: small, minor)

Introduced from Brazil (*Salvinia molesta*), Central and South America (*Salvinia minima*)
Found throughout the southern US

Introduction and spread

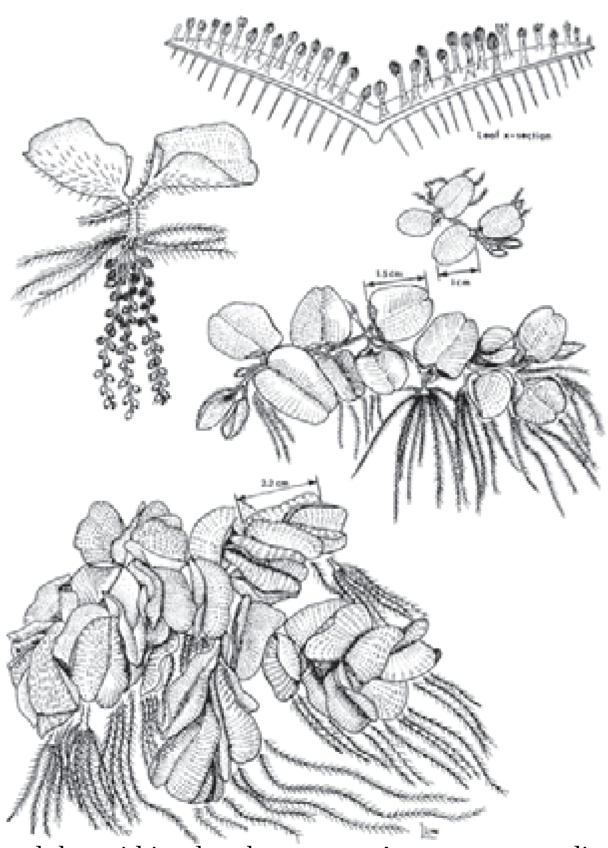
Water ferns in the genus *Salvinia* are members of the Salviniaceae family. There are 12 species of *Salvinia* reported worldwide, seven of which originate from the New World tropics (the Western Hemisphere). None of the *Salvinia*

species are native to North America, but two species – *Salvinia minima* and *Salvinia molesta* – have been introduced from South America and are currently established in the US. Both species were likely introduced into the US through the nursery trade as ornamental plants for water gardens or through the aquarium plant industry.

Salvinia molesta, commonly known as giant salvinia, is native to southeastern Brazil and was first found outside its native range in Sri Lanka in 1939. Giant salvinia quickly became a widespread weed problem in Sri Lanka, infesting rice paddies, reducing flows in irrigation channels and blocking navigation in transportation canals. Today, giant salvinia is considered one of the world's worst weeds and has become established in over 20 countries including Africa, India, Indonesia, Malaysia, Singapore, Papua New Guinea, Australia, New Zealand, Fiji, Cuba, Trinidad, Borneo, Columbia, Guyana, Philippines, and the US Territories of Puerto Rico and the Virgin Islands. In 2013, giant salvinia was added to the list of 100 of the World's Worse Invasive Alien Species by the International Union for Conservation of Nature (http://www.iucngisd.org/gisd/100_wort.php).

The first report of giant salvinia outside of cultivation in the US occurred in 1995 when it was discovered in a small, private pond in South Carolina. Once identified, it was quickly eradicated from this site with the use of herbicides. Although this initial infestation was successfully eradicated, giant salvinia has since been reintroduced and has spread throughout the southern US. Significant infestations have been reported in more than 90 locations in more than 41 freshwater drainage areas of 15 states (Alabama, Arizona, Arkansas, California, Georgia, Hawaii, Florida, Louisiana, Mississippi, Missouri, North and South Carolina, Oklahoma, Texas and Virginia) and the District of Columbia. Giant salvinia is currently listed as a Federal Noxious Weed by the US Department of Agriculture (www.aphis.usda.gov/ppq/weeds/), which prohibits its importation into the US as well as its transport across state lines. However, giant salvinia must be listed as a noxious species by individual states to prohibit sale and cultivation of the species within that state. Fourteen states currently list giant salvinia as a noxious weed or a prohibited weed/plant pest. Since this species is not currently designated as a noxious weed by all states, the expansion of giant salvinia will likely continue. Quarantine and sale of giant salvinia by the nursery industry has been difficult to enforce nationwide. In fact, a recent survey of mail-order catalogs and on-line commercial vendors for water garden enthusiasts revealed that giant salvinia was among the many noxious aquatic plants readily available for purchase over the internet.

Salvinia minima, hereafter referred to as common salvinia, is native to Central and South America. Outside its native range it has established in Bermuda, Puerto Rico, Spain and North America. Common salvinia was first reported in the US in 1928 along the St. John's River in Florida. The source of this first introduction to a natural area was likely the result of an unintentional release from a grower whose cultivation ponds had flooded. Since then, populations have been recorded in more than 80 freshwater drainage areas across the US. According to the most recent distribution records reported by the USGS Nonindigenous Aquatic Species Database, common salvinia is found in 17 states including Alabama, Arkansas, California, Florida, Georgia, Idaho, Louisiana, Maryland, Massachusetts, Mississippi, Missouri, New Mexico, New York, Ohio, Oklahoma, South Carolina and Texas. Similar to giant salvinia, common salvinia is widely available through the water garden trade. Although it continues to infest new regions, common salvinia is not listed as a Federal Noxious Weed; however, it is currently listed as a prohibited plant in Florida and a noxious weed/plant in North Carolina and Texas.



Description of the species

Common and giant salvinia are free-floating aquatic ferns with a horizontal stem or rhizome that floats at or just below the water surface. A pair of floating leaves or fronds (leaves of ferns are referred to as "fronds") are produced at each node along the rhizome. Fronds are bright green in color, oval in shape, possess a central midrib and are covered with numerous stiff, white hairs. It is thought that the function of these leaf hairs is to repel water and thus aid in plant buoyancy. An easy way to distinguish giant salvinia from common salvinia is by the shape of the hairs on the upper surface of floating fronds. The hairs on the fronds of giant salvinia form cage-like structures at the tip that resemble an eggbeater or kitchen whisk, whereas the hairs on common salvinia fronds are open at the tip and have a fringed appearance.

Common and giant salvinia lack true "roots" but possess delicate, finely-dissected submersed fronds. Submersed fronds are brown and resemble roots and serve a similar function by absorbing nutrients from the water. Sporocarps (structures that hold the fern's spores) are borne in chains or clusters on submersed stalks but do not bear fertile spores. Sporocarps are not found at all plant nodes but often develop and are more abundant later in the growing season or when nutrient conditions are poor.

Common salvinia

Giant salvinia

Both giant and common salvinia favor stagnant or slow-moving water habitats of lakes, ponds, rivers, streams, oxbows, ditches, canals, swamps, marshes and rice fields. Under favorable growing conditions, both species can form dense, expansive plant mats that can completely cover the water surface. Optimal growing conditions include full sunlight and warm (75 to 85 °F), nutrient-rich waters with a pH of 6 to 7.5. Upper and lower temperature thresholds for growth are about 95 and 50 °F, respectively. Both giant and common salvinia have a low tolerance to salinity and cannot survive in brackish or marine environments.

Reproduction

Giant and common salvinia are ferns, so they do not produce flowers or seed. As mentioned above, both species produce sporocarps that may contain spores but the spores are not thought to be viable. As a result, giant and common salvinia reproduce solely by vegetative means through fragmentation or the production of new plants from lateral and terminal buds. Stems may have as many as 5 buds per node and each bud is capable of developing new fronds. In addition, horizontal stems or rhizomes break apart very easily and produce fragments that disperse and develop into mature individual plants.

An individual giant salvinia can double in size in as little as 5 to 7 days when conditions are favorable. Some reports have calculated that a single giant salvinia plant can multiply to cover 40 square miles in 3 months under optimal growing conditions. With such an explosive growth rate, giant salvinia can quickly cover lakes and rivers, forming vegetative mats up to 3 feet thick. Common salvinia also has a rapid growth rate and can form dense mats, but is usually less aggressive than giant salvinia.

The major means of dispersal within and among lakes for giant and common salvinia is vegetative spread by fragmentation. Plant populations expand laterally within a lake through rhizome and lateral bud growth, whereas long distance dispersal is mostly the result of fragmentation. Plants easily adhere to boats, trailers, motors and other amphibious vehicles and can be transported to new locations. Animals (livestock, turtles, wading birds and waterfowl) may also contribute to the spread and dispersal of salvinia.

Problems associated with giant and common salvinia

Both giant and common salvinia can alter aquatic ecosystems in many ways. Dense growths can form a physical barrier on the water surface and hinder recreational activities such as boating, swimming, fishing and water skiing. Vegetative mats of salvinia can also impede navigation, impair flood control, limit irrigation, clog water intakes, decrease waterfront property values and cause problems in rice, catfish and crawfish production systems. Occasionally, other plant species (including grasses and small trees) will colonize or grow on mats of giant salvinia and create massive floating islands that can trap sediments and cause waterbodies to fill in over time.

Ecologically, extensive salvinia mats restrict light penetration and impede gas exchange between the water and atmosphere. Limiting light availability reduces photosynthesis of submersed aquatic plant communities and reduces water temperature. Low dissolved oxygen levels in the water are detrimental to fishes and other aquatic organisms and promote the accumulation of organic matter as microbial degradation is reduced. Changes in water quality can significantly impact the health of aquatic habitats and often result in declines in number and diversity of plant, invertebrate and animal communities. The loss of open water habitat also reduces the use of these areas by migrating waterfowl and wading birds (Sections 1.3 and 1.4).

Public health issues are also of concern. Both species of salvinia provide breeding habitats for mosquitoes and associated mosquito-borne illnesses (e.g., West Nile virus, malaria, encephalitis – Section 1.5). In Sri Lanka, it was reported that giant salvinia served as an important host plant and breeding habitat for mosquitoes which transmit filariasis (elephantiasis). Increases in the occurrence of schistosomiasis (which is transmitted by snails) have also been linked with large infestations of giant salvinia in developing countries.

Management options

Giant and common salvinia can be managed using herbicides, biocontrol agents, manual or mechanical harvesting, water level manipulation or a combination of these methods. Selecting the best management strategy depends on site-specific management goals and objectives, site characteristics, size and density of the infestation, proximity to sensitive plant or animal species, water body uses and budget constraints. The key to successfully managing giant and common salvinia is to recognize the problem early when infestations are small and can be easily contained. Small mats of salvinia are easily broken up by wind and wave action and are spread throughout large bodies of water. Once giant or common salvinia become well established and cover large areas, management becomes more difficult, time consuming and costly and may require multiple applications of a treatment method over a number of years to achieve maintenance control.

Herbicides (Section 3.7.1) can provide effective short and/or long-term control of giant and common salvinia depending on the choice of product and the method and frequency of application. Of the herbicides currently registered by the US Environmental Protection Agency for use in aquatic sites, ten provide good (> 75%) to excellent (> 90%) control of giant or common salvinia. The most widely used herbicides against these weed species include diquat, glyphosate, flumioxazin and carfentrazone-ethyl. Glyphosate applied in combination with diquat (plus appropriate surfactants; Section 3.7.3) is currently the most frequently used treatment for the management of giant salvinia in the US.

Diquat, flumioxazin, carfentrazone-ethyl and chelated copper formulations are non-selective contact herbicides that are typically applied as foliar sprays. Injury symptoms (severe leaf browning) are visible one day following application and plant death occurs within 3 to 4 days of treatment. Contact herbicides are fast-acting but have little or no movement inside plant tissues, so only plant tissues that come into contact with the herbicide are affected. Glyphosate is a non-selective, systemic herbicide that is applied to foliage, absorbed through the leaves and moves throughout the plant. Injury symptoms (leaf yellowing and browning) appear seven days after glyphosate application and plant death occurs by 28 days after treatment.

Other systemic herbicides that are effective, but slower-acting and used to a lesser extent against these two salvinia species, include imazamox, fluridone, penoxsulam, topramezone and bispyribac-sodium. Imazamox is effective on common salvinia but shows little or no activity on giant salvinia. Both species are susceptible to penoxsulam, bispyribac-sodium and fluridone. These herbicides require long contact times (60 to 90 days) to achieve control of salvinia, whereas imazamox has a shorter contact time requirement (7 days). Contact time refers to the length of time the target plant must be in contact with or exposed to a lethal dose of herbicide to achieve control. If contact time is not maintained because of water exchange or other factors that can cause dilution, plant control will be reduced. Imazamox and penoxsulam can be applied as a foliar spray or as a submersed application to the water column, whereas fluridone is effective only as an in-water treatment. Although in-water herbicide applications can be effective for treating these floating weed species, this method may not be feasible for sites where high water exchange or flow affect herbicide contact time and may be prohibitively expensive in larger systems.

Giant and common salvinia can be difficult to manage using herbicides because they are small floating plants that produce dense stands with plants layered on top of one another. This layering of plants presents a challenge when applying herbicides to foliage because plants in lower layers of the mats are protected from herbicides by plants in the upper layers of the mats. If plants are dense and a thick vegetative mat has formed, multiple applications will be required to achieve successful long-term control. In addition, giant and common salvinia can survive short dewatering or drawdown events and can persist on moist soils; therefore, spraying shoreline areas in addition to plants on the water surface is important to prevent re-infestation via surviving plant material. Long-term management with herbicides requires follow-up monitoring to spot-spray any plant material that survived the initial application. As a good management practice, herbicides should be routinely rotated and/or combined with other control strategies to minimize the potential development of herbicide resistance (Section 3.7.2).

Several insects have been investigated as biological control agents (Section 3.6.1) against salvinia species, but the salvinia weevil (*Cyrtobagous salviniae*) is recognized throughout the world as the insect of choice for management of giant and common salvinia. This insect feeds and reproduces only on plants in the Salviniaceae family. The salvinia weevil is a small (less than 1/16 inch long) black weevil that, like salvinia, is native to South America. Adults feed on floating fronds and rhizomes but prefer newly formed buds. The larvae of the salvinia weevil are white, 1/8 inch long and feed within the floating and submersed fronds, rhizomes and buds. Feeding by the larvae is often more destructive than that of adults. The combined feeding action of adults and larvae can be devastating and can impact field populations of giant and common salvinia in several months as opposed to the longer periods of time required by other insect biocontrol agents. Attacked plants turn brown in small patches that merge together until the whole colony loses structural integrity, becomes waterlogged and sinks. Although never intentionally released, the salvinia weevil was first detected in Florida in 1960, where it is now widespread and feeds primarily on common salvinia. Initial attempts to release weevils collected from Florida to manage giant salvinia in Texas and Louisiana were ineffective. This prompted researchers to seek permission from the Technical Advisory Group and the USDA-APHIS-PPQ (US Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine – see Section 3.6), to release a strain of the salvinia weevil from Australia which was highly effective in overseas applications. Permission was granted in 2001 and the Australian weevils were released in east Texas and western Louisiana only. The weevils, have become established in some localized sites and are beginning to impact giant salvinia populations. However, recent evidence suggests that *C. salviniae* fail to overwinter in temperate locales in Texas and Louisiana. The inability of *C. salviniae* to oviposit (lay eggs) and for larvae to develop below water temperatures of 66 °F and 63 °F, respectively, indicates the importance of water temperature as a limiting factor for the successful establishment of this biological control agent. Following cold winters, additional releases of *C. salviniae* will be required to reestablish the insect.

The potential utility of fungi as a biocontrol agent against giant salvinia has been investigated. Isolates of the fungal pathogens *Myrothecium roridum* and *M. verrucaria* have been evaluated as bioherbicides against several invasive weed species, including giant salvinia. While some isolates were shown to be virulent against salvinia, to date, plant pathogens have not been developed as biopesticides for use against this species.

Herbivorous fish such as triploid grass carp (Section 3.6.2) and tilapia (*Oreochromis* sp.) have been evaluated as possible biocontrol agents against salvinia with limited success. Laboratory feeding studies showed that while tilapia will consume giant salvinia, it is not their preferred food if other food sources are available. Other studies have shown that salvinia provides little nutritional benefit to herbivorous fishes.

The effectiveness of mechanical methods (Section 3.5) or manual removal (Section 3.4) is limited but may be useful in the early stages of an infestation or when a localized population is found on a small water body. If mechanical harvesting methods are employed, plant material must be properly disposed of in upland areas where the potential for contamination of other water bodies is minimized. Mechanical removal is not economically feasible once giant or common salvinia is well established and covers large areas. However, combining mechanical removal with herbicide applications can be an effective integrated weed management strategy. For example, in 2003, the Hawaii Department of Agriculture was successful in controlling 300 acres of giant salvinia on Lake Wilson on Oahu using multiple applications of the herbicide glyphosate combined with mechanical removal techniques. Excavated plant material was safely disposed of in nearby pineapple fields.

Other management options (Section 3.4). Floating booms have been used to contain and limit the spread of giant and common salvinia in some systems but are generally only utilized to confine plants to one location while other management strategies such as herbicides or weevils are deployed. Drawdowns can be a low-cost, effective management approach in some situations where water levels can be manipulated. However, dewatering must occur over a long period of time to allow plants to become stranded on dry land where they will desiccate and/or be exposed to freezing temperatures. Plant material can remain viable for several months if stranded shoreline mats are dense and underlying moisture is present. Decaying plant material along shorelines can be unsightly and plant fragments can easily be blown back into the system.

Summary

Giant and common salvinia are fast-growing, mat-forming aquatic ferns that can quickly cover the water surface of lakes, rivers and other wetland habitats. They are aggressive competitors that reproduce only by vegetative means. The plants can tolerate a wide range of growing conditions but prefer warm, nutrient-rich waters and full sunlight. Giant and common salvinia prefer freshwater environments and will not colonize saline or brackish waters. Once established, herbicides can be used to effectively manage these plants; however, multiple applications, follow-up monitoring and spot treatments may be required to maintain long-term control. Introducing insect biocontrol agents such as the salvinia weevil can be effective for maintenance control in some systems. The salvinia weevil has been especially successful in Florida for keeping common salvinia populations in check. Preventing the spread of this plant through citizen watch programs, boat launch surveillance and enforcement and compliance with laws to prevent the cultivation, sale and transport of these species will be important for containing and minimizing further spread of giant and common salvinia in the US.

Photo and illustration credits:

Page 79: Giant salvinia at Lake Wilson, Oahu; Linda Nelson, USACE ERDC
Page 80: Line drawing; University of Florida Center for Aquatic and Invasive Plants
Page 81 upper: Common salvinia; Ted Center, bugwood.org
Page 81 lower: Giant salvinia; Mic Julien, bugwood.org
Page 83: *Cyrtobagous salviniae* on giant salvinia frond; Scott Bauer, bugwood.org

2.14 Duckweed and Watermeal: The World's Smallest Flowering Plants

Tyler Koschnick: SePRO Corporation, Carmel IN; tylerk@sepro.com
Rob Richardson: North Carolina State University, Raleigh NC; rob_richardson@ncsu.edu
Ben Willis: SePRO Research and Technology Campus, Whitakers NC; ben.willis@sepro.com

Duckweed species can grow so densely on water surfaces that they appear as finely groomed turf. They are considered the world's smallest flowering plants. To put their size and numbers in perspective, watermeal is approximately the size of a sugar crystal or a grain of salt, which translates to 5 to 10 billion plants per acre.

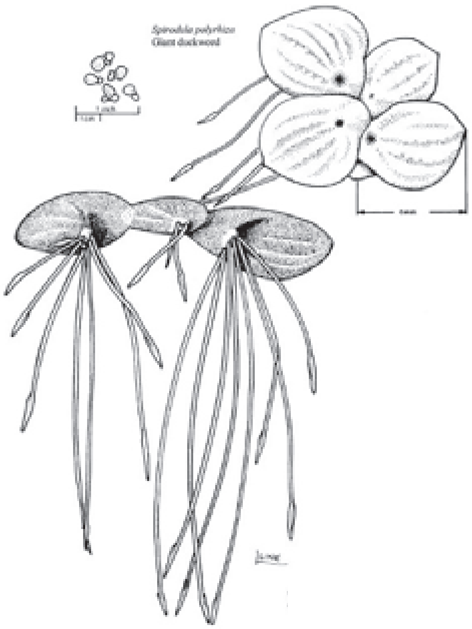
Introduction and spread

Duckweeds represent five genera of small floating aquatic plants in the *Araceae* subfamily *Lemnoideae* (although until recently duckweeds were considered members of the *Lemnaceae* or duckweed family). The duckweeds (*Landoltia*, *Lemna* and *Spirodela*), watermeal (*Wolffia*) and bogmat (*Wolffiella*) genera include more than 35 species worldwide; in this chapter, the term "duckweed" will refer to all members of these five genera. Multiple species are native to North America, such as *Spirodela polyrrhiza* (giant duckweed), *Lemna minor* (common duckweed), *Lemna minuta* (least duckweed) and *Lemna gibba* (swollen duckweed), but some species found in the US – including the Australian or Southeast Asian native dotted duckweed (*Landoltia punctata*) – are introduced. Duckweed is widespread in distribution and is found on every continent except Antarctica. Some species, like *Lemna minor*, are native to multiple continents. Growth rates are extremely high and populations can double in size in 1 to 3 days under optimal conditions. The diminutive size of duckweed allows plants to easily "hitchhike" on water currents, waterfowl and watercraft, which contributes to its spread.

Although duckweeds are often a nuisance in backyard ponds, the plants are valued and used extensively for applied and basic plant science research. Duckweeds have many potential uses, including biofuel production and as a food source (duckweed reportedly tastes like spinach and is high in protein and vitamins). Duckweeds have also been used as bioremediation agents to remove waterborne nutrients and contaminants. These species can improve water quality in natural systems such as lakes and can reduce nitrogen, phosphorus and metal contamination in commercial waters such as swine-based effluent ponds before they are discharged to other waters, although this could accidentally introduce duckweeds to downstream systems.

Description of species

Duckweeds are monocotyledons and can be distinguished from other floating plants by their small size, which ranges from around a 1/25 of an inch to less than an inch. Duckweeds have the distinction of being the world's smallest flowering plants and some species, especially bogmat and watermeal, are commonly confused with algae. Another floating aquatic plant that could be confused with duckweed is the native mosquito fern (*Azolla caroliniana*). Mosquito ferns are diminutive like duckweeds but are branched instead of round and plants are often red, particularly when grown in full sun.



Duckweed species can be separated based on: 1) frond size, number and shape, and 2) root structure or lack thereof. Fronds are leaf-like structures and may be modified stem or leaf systems that absorb nutrients from the water column. The function of the modified root structure is not well understood (although the roots may help the plant stay in an upright position), and roots are lacking in the genera *Wolffiella* and *Wolffia*.

The largest duckweeds are up to one inch in diameter and belong to the genus *Spirodela*. Plants in this genus are also the most structurally complex of the duckweeds and have flowers and many roots per frond. Duckweeds in the genus *Landoltia* are similar to *Spirodela* duckweeds, but are smaller (around one third the size), have fewer roots (from several to one per frond) and usually lack the distinctive dot on the frond surface that is characteristic of *Spirodela* species. Duckweeds in the genus *Lemna*, which have one to several fronds and a single root per frond, are smaller than members of the genus *Landoltia*. Plants in the genera *Wolffiella* and *Wolffia* are the smallest of the duckweeds and have the least complex structure (no roots, simplified flowers). These genera can be identified by their fronds, which are long and spindly in *Wolffiella* and oval in *Wolffia*. Although it is fairly easy to distinguish among the duckweed genera, it is much more challenging to identify species within each genus, particularly in the *Lemna* duckweeds.

Duckweeds are typically found in still, nutrient-rich waters, and populations or colonies of tens of thousands of individual plants can thrive in small pools of water or ditches. Some duckweed species can survive cold (but not freezing) temperatures and increases in salinity can stimulate growth, although excess salinity can inhibit growth or kill plants. Duckweeds can provide habitat for many aquatic organisms such as insects and frogs and can be an important food source for wildlife, including fish and birds (hence the name “duckweed”).

Reproduction
Duckweeds are very productive and might very well be among the fastest growing plants. Despite being the smallest angiosperms, flowers are rarely seen due to size and blooming frequency. The small fruit produced is called a utricle. Duckweed primarily reproduces through asexual vegetative budding where each frond produces a new plant. This mode of growth can allow duckweed to quickly cover ponds and lakes with an extremely short doubling time. Multiple species can produce seeds and turions (or buds) for overwintering; one seed is produced per frond. Turions are modified structures that sink to the bottom of lakes where they overwinter, but not all duckweed species produce them. Seed production is a particularly important adaptation that allows survival of droughts. Seeds are reported to have extremely low survival (if any) after exposure to freezing conditions, which limits overwintering capabilities. However, duckweed seeds and turions are adapted to sink to the bottom of water bodies to escape freezing for insuring a viable propagule bank for growth in warmer conditions.

Problems associated with duckweed
Similar to filamentous algae, duckweed can form dense surface mats that are several layers thick and may include mixtures of different species. However, duckweed’s ability to decrease light penetration and intensity and to consume nutrients can actually inhibit algal growth. Dissolved oxygen concentrations below duckweed mats are often low, which can influence the type and abundance of invertebrate and fish populations. Duckweed mats can also reduce aesthetics and recreational uses of water resources because their excessive growth covers the surface of the water. Duckweed usually causes problems in smaller bodies of water such as backyard ponds, canals, wetlands and other static sites. However, it has also created significant issues on some very large lakes, including Lake Maracaibo in Venezuela (South America’s largest lake).

Management options

Duckweeds can present an extreme challenge to resource managers. Control methods provide only temporary relief; unless every plant is successfully managed, colonies will rapidly re-form because duckweeds reproduce so quickly. In addition, duckweed can survive on mud flats and wet shorelines, which allows them to escape management efforts. These missed plants can quickly re-infest a site once they are flushed back in to the water by wave action or rising water levels. In addition, upstream sources that host colonies of duckweed can also be a source of new introductions.


Spirodela


Landoltia


Lemna


Wolffiella


Wolffia

Floating booms and suction devices can be used to remove duckweed, and rakes can be used when wind and currents cause colonies to accumulate near banks or in isolated small areas (Sections 3.4 and 3.5). However, mechanical harvesting is typically limited to smaller (less than 1/2 acre) water bodies. Dyes do not provide control of duckweed and may actually promote growth of colonies by reducing algal competition. Aeration can relieve the low dissolved oxygen levels associated with large duckweed populations, thus improving fish habitat, but do not affect plant growth. Grass carp (Section 3.6.2) have been used to manage small infestations of duckweed, although high stocking rates (50 to 75 per acre) of small fish (4 to 6 inches) are needed to have an impact. It is important to remember that small grass carp are very susceptible to predation, so most stocking recommendations specify grass carp that are at least 10 to 12 inch long to reduce predation. However, grass carp that are this large have lost the ability to strain small plants from the water and have little utility for duckweed control.

Chemical control (Section 3.7.1) is the predominant method used to manage duckweed, but different species of duckweeds have differing susceptibilities to herbicides. For example, *Lemna* duckweeds are generally considered easier to control and more susceptible to herbicides than *Wolffia* (watermeal), which are the most difficult species of duckweeds to control. Since these plants often co-exist, it is possible to successfully control one species (*Lemna* duckweed) without causing significant damage to the other (watermeal). Therefore, proper identification of the genera targeted for management is very important. General guidelines for managing *Lemnoideae* species with herbicides are outlined below; however, it is important to remember that effectiveness of control methods are species-dependent and can vary.

There are multiple herbicides that may be used to control duckweed; these are generally separated into slower acting, systemic and faster acting, contact herbicides. These herbicides may be applied in-water (absorbed by the plant primarily from the water column) or to plant foliage (applied directly to the surface of the plant). In-water systemic herbicides are used to manage duckweed when populations cover large areas (or the entire surface) of a water body. These products are relatively easy to apply and, when effective, usually result in long-term control. In-water systemic herbicides can be applied to the surface of the water or can be injected directly into the water column and need to maintain contact with the plant for an extended period of time. Contact with every individual plant during the application is not required because herbicides applied in-water diffuse through the water column. These herbicides are slower-acting, so large infestations can be treated without negatively affecting dissolved oxygen levels because plant death occurs over an extended period. Fluridone has historically been the most commonly used in-water herbicide for duckweed control, but penoxsulam and bispyribac-sodium are also labeled to control duckweed. The foliar-applied systemic herbicides glyphosate and imazapyr are unlikely to provide long-term control of duckweed because these products become ineffective once they enter the water column and individual plant coverage is insufficient for control.

Depending on conditions and the scale of application, contact herbicides such as diquat and flumioxazin may provide effective control of duckweed. (Note: *Wolffia* duckweeds are generally tolerant of diquat, so foliar applications of diquat alone are not recommended for control of *Wolffia* duckweeds. Foliar applications are also not recommended for *Wolffiella*, although this species is rarely targeted for control.) Other contact herbicides such as chelated coppers are labeled for duckweed control but are not commonly used unless local conditions or water-use restrictions limit other options. Contact herbicides are faster-acting with shorter half-lives in water, so they must be applied to the entire surface area of the duckweed population or as an in-water application to the entire water body, but close to the surface. Surfactants (Section 3.7.3) should not be used when applying faster acting herbicides as a foliar treatment to duckweed because these products can cause plants to “sink”, which washes the herbicide off the leaf surface and reduces efficacy. Foliar treatments that are applied by boat inevitably result in some wash-off as well. Care should be taken to avoid sinking or wash-off during the application process because good coverage is critical when using foliar herbicides. Also, if duckweed colonies are extremely dense, mats might be several layers thick and a foliar application might kill only the plants on the surface of the mat. In this situation, plants in lower layers of the mat are unaffected and can quickly re-colonize the surface of the water. As a result, foliar herbicide applications must often be repeated to control remaining plants that escaped direct exposure to the herbicide during the initial application.

Because contact herbicides act quickly, these products are typically applied to only part of the water body at one time; this helps to avoid the major reduction in dissolved oxygen that can occur when large populations of plants are killed. Some contact herbicides prohibit treating more than one-third to one-half of a water body if dense vegetation is present but allow application of the product to untreated areas 10 to 14 days after the initial application. Contact herbicides should be used as early in the growing season as possible – before peak plant growth and while water temperatures are cooler – to help reduce oxygen depletion.

The first documented case of herbicide resistance (Section 3.7.2) in floating aquatic plants occurred in *Landoltia punctata*; however, this species’ resistance to diquat was reduced when copper was applied in combination with diquat. Using a combination of systemic and contact herbicides (for example, fluridone plus flumioxazin) could improve efficacy and provide longer-term control at lower rates than either product would when applied individually.

Summary

Members of the five duckweed genera are widespread and occur on almost every continent. Despite their diminutive size, these plants can form dense multi-species colonies on the surface of the water, which decreases water quality and impedes recreational and other water resource uses. Duckweeds commonly cause issues in small ponds and canals but are sometimes problematic in larger systems. Mechanical and biological methods are sometimes used for management, but their use is often limited. However, there are several options for chemical control that can be used to manage nuisance colonies of duckweed.

Photo and illustration credits:

Page 85: Duckweed infestation; Tyler Koschnick, SePRO Corporation
Page 86: Line drawing; University of Florida Center for Aquatic and Invasive Plants
Page 87: Duckweed montage; Ben Willis, SePRO Corporation

2.15 Phragmites: Common Reed

Candice M. Prince: University of Florida, Gainesville FL; cprince14@ufl.edu
Jack M. Whetstone: Clemson University, Georgetown SC (emeritus); jwhtstn@clemson.edu

Phragmites australis (Cav.) Trin. ex Steud; emergent plant in the Poaceae (grass family)
Derived from *phragma* (Greek: fence) and *australis* (Latin: southern): "southern plant with fence-like growth"

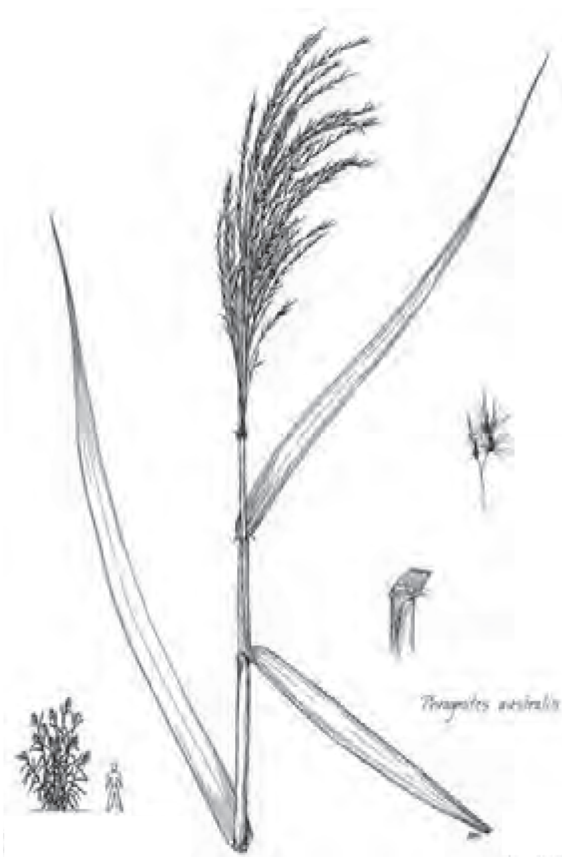
Invasive Eurasian lineage probably introduced from Europe to the northeastern Atlantic Coast (early 1800's)
Gulf Coast lineage (*Phragmites australis* var. *berlandieri* (E. Fourn.) C.F. Reed) in the southeastern United States (unclear if native or exotic)
Native lineage (*Phragmites australis* subsp. *americanus* Saltonstall, Peterson & Soreng) historically widespread throughout North America, except for the southeastern United States

Introduction and spread

Phragmites (also called common reed) is a tall wetland grass that grows from thick white rhizomes (underground stems) in areas with fresh to brackish water. It is one of the most common flowering plants in the world and is found on every

continent except for Antarctica. Phragmites occurs in every US state except Alaska and in every Canadian province except Nunavut and Yukon. There are three lineages of phragmites in North America – a native lineage (*Phragmites australis* subsp. *americanus*), a Gulf Coast lineage whose origins are unclear (*Phragmites australis* var. *berlandieri*) and an invasive Eurasian lineage.

Native phragmites (*P. australis* subsp. *americanus*) was historically found throughout North America except for the southeastern US. It has been replaced by the Eurasian type in much of its historic range (especially in the Northeast) but can still be found in the Midwest and western states. The Gulf Coast type of phragmites can be found from Florida to California in the southern US. It is unknown whether it is native or was introduced to the Gulf Coast region, but it is not considered a problem species in many areas. However, the Gulf Coast type may grow aggressively in the southwestern US and in some disturbed wetlands in the Southeast, where it can completely replace other vegetation if not properly managed. The Eurasian type is much more invasive than the native and Gulf Coast types and is able to outcompete them for space and resources. It was introduced to the northeastern US in the early 1800s in ship ballast. The Eurasian type is now the dominant type of phragmites along the Atlantic Coast and in the Great Lakes area and is continuing to increase in western and southeastern states. This type of phragmites is very aggressive and has been called an "ecosystem engineer" because it changes the areas it invades and replaces other vegetation. Large monotypic (single-variety) stands of Eurasian phragmites are associated with decreased plant diversity. In addition, soil properties, sedimentation rates and wildlife habitat may be altered when marshes are converted from diverse plant communities to dense, monotypic stands of phragmites.



Phragmites can be found in many different habitats. It is most common in freshwater marshes, salt marshes and on the edges of lakes, irrigation ditches and other waterways. The species can tolerate seasonal drought as well as frequent, prolonged flooding. Phragmites grows best in sites with fresh to brackish water (0 to 5 parts per thousand salinity) but it is highly salt tolerant and can reportedly survive in areas that are as saline as full strength ocean water (35 parts per thousand). It establishes and grows well on disturbed sites such as roadsides and is often considered a weedy or nuisance species.

Description of the species

Phragmites is a perennial grass that can reach up to 20 feet in height, but is typically 10 to 12 feet tall. Leaves are 4 to 20 inches long, 0.4 to 2 inches wide and are blue-green to green in color. Leaves are hairless with rough margins and are arranged in an alternate manner on the stems. The ligule (outgrowth on the upper leaf surface where the leaf blade meets the leaf sheath) is a fringe of long hairs. Stems are stout, erect and hollow and will die back during the winter. Phragmites flowers in late summer through fall and produces flowers in large, light brown panicles (1 to 2 feet in length) that form silky hairs at maturity.

The different lineages of phragmites are very difficult to tell apart in the field and genetic testing is required for identification. However, there are a few traits that may help distinguish between the three major lineages. The native type often grows in mixed stands with other species, whereas the Gulf Coast and Eurasian types form monocultures with high stem densities. Stems of the native and Gulf Coast types are smooth and shiny, while stems of the Eurasian type are dull and have ridges. The flowers of the native and Eurasian types are both upright, but are much more dense in the Eurasian type. In contrast, the flowers of the Gulf Coast type are drooping and more open in appearance.

Reproduction

Phragmites spreads across long distances primarily through seeds, although transportation of rhizome fragments can also play a major role in dispersal. Seeds are spread by wind and water and viability is variable. Local spread of phragmites is mainly through rhizomes and stolons (creeping horizontal plant stems). Rhizomes have been found buried up to 30 feet deep in the substrate and can spread laterally over 15 inches per year, while stolons can grow more than 4 inches per day and may reach 40 feet in length in a growing season.

Problems associated with phragmites

Invasive phragmites forms large monotypic stands that are virtually impenetrable. These stands replace diverse native plant communities and reduce ecosystem productivity. The Eurasian type of phragmites alters soil properties and sediment accumulation rates and reduces wildlife habitat. However, phragmites does provide minor shade, nesting and cover habitat for mammals and fishes. Phragmites also provides food and sites for nesting, roosting and hunting for a wide variety of bird species, including ducks (Section 1.3). Waterfowl, pheasants and rabbits use the margins of stands of phragmites as cover to hide from predators.

Habitat use by fish (Section 1.2), crustaceans and other aquatic invertebrates can be affected by dense growth of phragmites. For example, small fish and crustaceans prefer habitats with low growing, less dense stands of native smooth cordgrass (*Spartina alterniflora*) to those with infestations of phragmites and populations of aquatic invertebrates are generally highest in areas with other native vegetation such as cattail (*Typha* sp.). Also, several studies report that marshes dominated by phragmites provide less suitable habitat for larvae and small juvenile forms of mud minnow.

Management options

As with any invasive plant, preventing establishment of phragmites is the best available option. This can be challenging due to the similar appearance of Eurasian, Gulf Coast and native phragmites, so you should consult an expert to help identify phragmites populations that are growing aggressively. Eurasian phragmites expanded from the Northeast by hitchhiking on equipment used in ditching, drainage and dredging operations. Therefore, inspecting and cleaning equipment is important before moving equipment into new areas to prevent the dispersal of any aquatic invasive plants, but particularly invasive varieties of phragmites. In addition, maintaining populations of competitive native plant species such as black needlerush (*Juncus roemerianus*) around phragmites stands may help limit vegetative spread.

Chemical control methods (Section 3.7.1) are the most common and effective method of controlling phragmites. There are a number of herbicides currently labeled for control of phragmites in aquatic habitats, including glyphosate, imazapyr, imazamox and triclopyr. Glyphosate and imazapyr are generally the most effective products for phragmites control, although the criteria for herbicide selection are site-specific and dependent on environmental conditions, growth stage of the plant, presence of desirable nontarget plant species in the area and alternate uses of the water such as drinking and irrigation.

Herbicides may be more effective if applied early in the growing season in areas where plants remain evergreen and grow throughout the year. However, early season treatments could result in winter regrowth, especially in areas such as the coastal Carolinas where it is not possible to completely dewater the soil before herbicide application. Thus, late summer or fall applications may be more effective in areas where the soil remains wet, phragmites goes dormant or where frost is probable. Phragmites often occurs in large, difficult to access areas, so aerial applications may offer the most efficient method for treatment (Section 3.7.4). Backpack sprayers can also be used for small infestations and spot treatments. Plants should be carefully sprayed to wet but runoff should be avoided and herbicide labels list more specific instructions for mixing and usage. Multiple yearly applications are often required for phragmites control and monitoring should be done to track regrowth.

Burning (Section 3.4) or mechanical removal (Section 3.5) of plants is not likely to provide adequate long-term control of phragmites. Burning may provide short-term control for small infestations but neither method can effectively reduce the extensive rhizome systems of large phragmites monocultures. However, integration of burning with flooding or herbicide application has been more successful; for example, one effective strategy is to burn, mow or cut the plants, then apply herbicide after plants begin to regrow. Flooding following a burn may also effectively control phragmites. Another strategy that may promote native plant recovery is to burn following herbicide application since this removes the leaf litter and dead thatch left behind by phragmites and can increase germination success of some native species.

Biological control (Section 3.6) of phragmites is complicated due to the presence of both native and invasive lineages in North America. No purposeful introductions of insects, pathogens or diseases for phragmites control have been attempted to date. Some reports suggest that immature plants are readily eaten by goats, cattle and horses but mature plants not considered to be high-value or highly palatable food for livestock. However, livestock grazing can be useful as part of an integrated strategy with other methods. In addition, two European shoot mining noctuid moths (*Archanara geminipuncta* and *A. neurica*) have recently been evaluated for phragmites control and were found to prefer the Eurasian type over the native type. Biological control may be a viable solution for phragmites management in North America in the coming years.

Summary

Phragmites is a widely distributed wetland species with three major lineages in the US: a native lineage, an invasive Eurasian lineage and a Gulf Coast lineage whose origins remain unclear. The Eurasian type has mostly replaced the native lineage in the northeast and has become established throughout the southeastern and midwestern states. The Gulf Coast type is problematic in western states and in disturbed wetlands in the Southeast. Invasive phragmites populations create large monotypic populations which are associated with decreased plant diversity and changes in soil properties, sedimentation rates, bird and fish habitat use and food webs. Phragmites management is made more challenging by the presence of the native type, which can be a desirable part of aquatic ecosystems and is similar in appearance to the Eurasian and Gulf Coast types. A variety of methods can be used to control invasive phragmites and greatest success is realized when multiple methods (such as burning followed by herbicide application) are employed in an integrated program.

Photo and illustration credits:

Page 89: Common reed; Candice Prince, University of Florida
Page 90: Line drawing; University of Florida Center for Aquatic and Invasive Plants
Page 91: Common reed; Ann Murray, University of Florida

2.16 Purple Loosestrife

Robert L. Johnson: Cornell University, Ithaca NY (retired); rlj5@cornell.edu

Lythrum salicaria L.; erect, emergent perennial herb in the Lythraceae (loosestrife) family
Derived from *lythrum* (Greek: blood) and *salicaria* (Latin: willow-like) “plant that stops blood and is willow-like”

Introduced from Europe to the east coast of North America in the early 1800s
Present in every state throughout the US, but not recorded in Florida, and found in all Canadian provinces

Introduction and spread

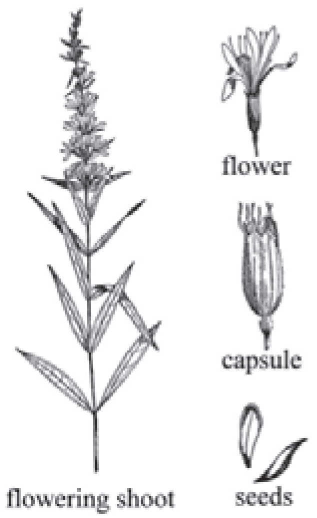
Lythrum salicaria L. (purple loosestrife) is often referred to as “the purple plague” in North America and is native to Europe and Asia. Purple loosestrife is an aggressive invasive plant deliberately introduced to the eastern coast of North America in the early 1800s. Settlers of the region valued the plant as an ornamental for perennial gardens and used the species as a medicinal herb to treat dysentery, diarrhea, bleeding and ulcers. The honey trade also increased regional seed propagation of the plant because it was a favored bee forage. In addition, European ships contributed to the spread of purple loosestrife by releasing ballast water and delivering shipments of wool that contained seeds of the species. By the 1830s, purple loosestrife had become established along the New England seaboard and the range of the species further expanded throughout New York State and the St. Lawrence River Valley through inland canals constructed in the late 1880s. As road systems expanded and commercial distribution of the plant by the nursery trade increased, purple loosestrife spread westward and southward invading every state and province of the US and Canada, while still not recorded in Florida. Purple loosestrife grows in most freshwater wetlands but also tolerates a wide range of environmental conditions and can spread to both tidal and non-tidal brackish waters.

Description of the species

Purple loosestrife is an erect, emergent perennial dicot herb with a dense, bushy appearance. The species tolerates a wide range of wetland environments and grows in habitats ranging from pastures with moist soil to sites with shallow water such as marshes and lakeshores. Established plants can tolerate a variety of soil conditions, including soils that are dry or permanently flooded and soils that are low in pH and nutrients. In addition, plants can grow in rock crevasses, on gravel, sand, clay or organic soils. Purple loosestrife can grow from four to ten feet in height and has a dense canopy of stems that emerge from its wide-topped crown. Each plant produces as many as 50 square, hard, red to purple stems that arise from a single root mass. Leaves are 1-1/2 to 4 inches long and 2/10 to 6/10 inch wide and are lance-shaped, stalk-less, heart-shaped or rounded at the base and borne in an opposite or whorled arrangement. Purple loosestrife produces flowers with magenta, purple, pink or white petals that are 4/10 to 8/10 inch long. The species blooms throughout most of the summer, which adds to its appeal as an ornamental plant and as a favorite of beekeepers. The reddish-brown seeds are very small (1/25 inch long) and are often produced during the first growing season. Purple loosestrife is often confused with several plants with spikes of purple flowers, including prairie blazing star (*Liatris pycnostachya*), blue vervain (*Verbena hastata*) and fireweed (*Chamaenerion angustifolium*). However, the species most closely resembles the native winged loosestrife (*Lythrum alatum*) and *Lythrum virgatum* L., a nonnative cultivated purple loosestrife. *L. virgatum* is very similar to purple loosestrife in appearance and formerly classified as a separate species, but considered by some to be a subspecies or variant form of purple loosestrife.

Reproduction

The extended flowering season of purple loosestrife typically lasts from June to September and allows each mature plant to produce more than 2 million seeds each year. Long-tongued insects, including bees and butterflies, serve as pollinators. Seed dispersal is often by water and seeds can hitchhike in mud that adheres to wildlife, livestock and people. Seed survival can be as high as 60 to 70%, which produces a sizeable seedbank in only a few years. Germination occurs in open, wet soils as temperatures increase in the spring, but seeds can remain dormant and viable for many years in the soil. In addition, submersed seeds can survive for up to 20 months in flooded conditions. Purple loosestrife readily colonizes newly disturbed areas because of its high production of viable seeds with multiple modes of dispersal. Disturbed areas with exposed soil are most vulnerable to invasion and rapid colonization by purple loosestrife because these sites provide ideal conditions for seed germination and usually lack native plants that compete with the weed for resources. Purple loosestrife spreads predominately via seed dispersal, but also by vegetative means through production of new shoots and roots from clipped, trampled or buried plants. Purple loosestrife's ability to reproduce via vegetative processes is an especially important consideration when contemplating adoption of management strategies. Mechanical or physical control efforts can inadvertently spread harvested plant fragments and create new infestation sites. In addition, disturbances in the form of changes in water levels from drought or a planned water drawdown provide ideal conditions for maximum seed germination and growth.



Problems associated with purple loosestrife

Purple loosestrife aggressively invades many types of wetlands, including freshwater wet meadows, tidal and non-tidal marshes, river and stream banks, pond edges, reservoirs and roadside ditches. The formation of dense, monotypic stands of purple loosestrife suppresses native plant species, decreases biodiversity and leads to a change in the wetland's community structure and hydrological functioning, while eliminating open water habitat in many locations. The United States historically has lost around 200,000 acres of wetlands every year due to invasions of purple loosestrife and has spent as much as $45 to $50 million per year on efforts to control the growth of this species. In addition to funds spent on control efforts, economic losses to agriculture can exceed millions of dollars annually when purple loosestrife invades irrigation systems. Also, the loss of entire crops of wild rice can occur when this species invades shallow lakes and bays dominated by wild rice, which results in great economic loss to local agricultural communities.

Purple loosestrife alters the physical makeup of a wetland but the species can change the chemical properties of the wetland as well. For example, the shedding of purple loosestrife leaves in the fall that rapidly decompose, allow a quick flushing of nutrients out of the wetland. In contrast, the vegetation of native species does not fully decompose until the following spring maintaining nutrients in the wetland throughout the fall and winter. This difference in the timing of nutrient release means that wetland decomposers have fewer nutrients available to subsidize peak population growth in the spring, which alters the structure of the food web. The effects of altered water chemistry extend to many of the fauna in aquatic ecosystems as well. For example, chemicals released during the decomposition of purple loosestrife leaves can slow the development of certain frog tadpoles, which decreases the frog's chance of surviving its first winter. Research at Cornell University suggests an underestimation of the threats to amphibians by nonnative plants. Their data indicate that organisms that breathe through gills (especially *Bufo americanus*, the American toad) are sensitive to the high concentration of tannins naturally produced during purple loosestrife decomposition.

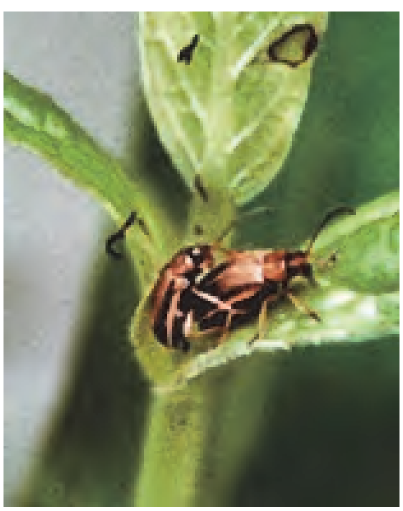
Purple loosestrife further affects the wildlife communities of wetlands through a variety of other means. The species is a very poor food source for herbivores and crowds out species that are more beneficial to the wetland food web. As a result, stands of purple loosestrife can jeopardize threatened and endangered plants and wildlife, especially in the northern US. For example, the bog turtle has lost extensive basking and breeding habitat due to the introduction of this aggressive plant. Purple loosestrife also displaces native plants such as cattail (*Typha* sp.) and bulrush (*Scirpus* sp., *Schoenoplectus* sp.), which provide high quality habitat to numerous nesting birds and aquatic furbearers. Wetland specialists such as the marsh wren, red-winged blackbird or least bittern (Section 1.4) prefer sturdy nesting sites such as cattail-dominated wetlands and are unable to utilize purple loosestrife for their nests. Also, muskrat, beaver and waterfowl prefer cattail marshes and are more able to utilize sites that native plants dominate compared to dense, monotypic populations of purple loosestrife.

A primary problem associated with purple loosestrife is its attractiveness. European immigrants to the US deliberately imported purple loosestrife as an ornamental plant in the 1800s and homeowners still actively plant the species today. Purple loosestrife may add a welcome burst of color to an otherwise dull private garden or pond, but the adaptability and aggressiveness of this plant can quickly wreak havoc on the unsuspecting homeowner's backyard. The sale or distribution of purple loosestrife is illegal in most states; however, nurseries, greenhouses and mail order sources sell the plant in many areas across the country and some seed mixes include the plant seeds. Consumers should always read seed package labels before purchasing in order to ensure that this aggressive nonnative plant is not in the mix.

Management options

The best way to stop an invasion of purple loosestrife is to be aware of pioneering plants and small isolated colonies. In these cases, hand removal of small, isolated stands is an effective preventative control method. The use of physical (Section 3.4) and mechanical (Section 3.5) control methods may provide annual control of low-density invasions and can include water level manipulation, hand removal, cutting and burning. These control methods should take place before seed production to avoid seed dispersal and contributions to the seed bank. It is also essential to remove roots from the soil since plants will regrow from broken roots or root fragments. Removal of flowering spikes will prevent seed formation and cutting or harvesting stems at the ground level will inhibit growth temporarily. While these methods temporarily halt growth, their use should be in conjunction with herbicides or biological control agents to provide longer-term management.

Annual applications of herbicides (Section 3.7.1) can be effective and can provide relatively successful season-long control of purple loosestrife stands but it is important to note that most states require application permits before using herbicides for management of purple loosestrife in wetlands or other aquatic locations. Control rates of greater than 90% can be realized with applications of the herbicides glyphosate and imazapyr; also, triclopyr (alone or mixed with 2,4-D) provides control for purple loosestrife. Single applications of registered herbicides generally do not provide satisfactory control of loosestrife for more than one season, but the use of imazapyr and glyphosate can result in multi-season control of purple loosestrife. Herbicides used to control purple loosestrife have very different selectivity spectrums for nontarget plants and application rate affects selectivity. When selecting an herbicide for management of purple loosestrife, it is important to consider potential negative impacts of the herbicide on the many important nontarget wetland species from overspray or exposure to high concentrations of herbicides needed to effectively control purple loosestrife.



The vast seedbank in the soil of established stands of purple loosestrife facilitates regrowth of the species after herbicides dissipate and are no longer effective. Therefore, the most effective long-term option for suppressing and controlling the growth of this invasive weed may be the use of biological control (Section 3.6). Research and evaluation of potential biological control agents for the North American purple loosestrife invasion identified European insects that showed promise as biocontrol agents. The USDA-APHIS approved four European insect species for introduction as classical biocontrol agents that have become well established (Section 3.6.1). These include two leaf-feeding beetles [*Galerucella calmariensis* L. and *G. pusilla* Duftschmidt (Coleoptera: Chrysomelidae)], a root-mining weevil [*Hylobius transversovittatus* Goeze (Coleoptera: Curculionidae)] and a flower-feeding weevil [*Nanophyes marmoratus* Goeze (Coleoptera: Curculionidae)]. Initial releases of the leaf-feeding beetles and the root-mining weevil into natural areas from New York to Oregon were experimental and early observations suggested that the leaf-feeding beetles occasionally feed on native plant species. However, this now appears to be of little consequence; both species have shown great success across the United States and Canada and are now becoming well-established.

The introduced European leaf-feeding beetles described above may be easily confused with native North American *Galerucella* species but the introduced beetles seriously affect purple loosestrife growth and seed production by feeding on the leaves and new shoot growth. The two introduced beetles are similar in appearance and share similar life history characteristics. Adults overwinter in leaf litter and emerge in the spring shortly after shoot growth begins. Peak dispersal of overwintered beetles occurs during the first few weeks of spring when new-generation beetles make dispersal flights shortly after emergence and can locate host patches greater than a half mile away within only a few days. Adults feed on shoot tips and females lay 2 to 10 eggs on the leaves and stems of purple loosestrife from May to July. Young larvae feed on developing leaf buds, while older larvae feed on all aboveground plant parts. Pupation by mature larvae takes place in the litter below the plant. Reports from several locations describe complete defoliation of large multi-acre stands of purple loosestrife, with local biomass reductions of greater than 95%. These positive results after years of introductions are extensive while sometimes localized, but have occurred in states ranging from the East to West Coasts and into the provinces of Canada.

Larvae of the introduced root-boring weevil hatch and feed on root tissue for one to two years depending on environmental conditions. Pupation occurs in the upper part of the root, with adults emerging between June and October. Adults then feed on foliage and stem tissue and can live for several years. The root-boring weevil can survive in all potential purple loosestrife habitats except for permanently flooded sites. Adults and larvae can survive extended submergence, depending on the temperature, but excessive flooding prevents access to plants by adults and eventually kills developing larvae. Feeding by adults has little effect on the plants, but as is typical, feeding by larvae can be very destructive to the rootstock.

Several states introduced the flower-eating weevil which is widespread in Europe and Asia, where it tolerates a wide range of environmental conditions. The flower-eating weevil greatly reduces seed production of purple loosestrife as larvae consume the flower and mature larvae form a pupation chamber at the bottom of the bud. Damaged buds do not flower with buds aborting, thus reducing purple loosestrife seed output. New-generation beetles appear mainly in August and feed on the remaining green leaves of purple loosestrife. Adults overwinter in leaf litter; development from egg to adult takes about 1 month and there is one generation per year.

Summary

The introduction of purple loosestrife into North America occurred in the early 1800s with the importation of wool containing seeds, as a favorite herb in flower gardens and from released ship ballast water. Unfortunately, this attractive plant has become one of North America's most widely dispersed and dominant nonnatives in habitats ranging from dry soils to inundated marsh areas or lakes. Stems can grow as tall as 10 feet and can form densities of up to 50 stems per plant, creating a canopy that limits light and space to native plants. Purple loosestrife causes problems in wetland ecosystems by forming dense monocultures, outcompeting native plants, altering hydrology and changing water chemistry, which all in turn affect native plant and animal communities. Purple loosestrife is an easily identified emergent plant, which facilitates hand removal and selective herbicide applications. Although limited, these methods can provide temporary control of small populations. Multiple control methods used in combination can effectively control populations, but biocontrol seems to provide the best long-term suppression of large dense stands of purple loosestrife. Fortunately, classical biocontrol agents appear to be able to successfully reduce populations of purple loosestrife throughout North America.

Photo and illustration credits:

Page 93: Purple loosestrife; Bernd Blossey
Page 94: Line drawing; adapted from Muenscher (1967)
Page 95: Mating pair of the leaf-feeding beetle *Galerucella calmariensis*; Bernd Blossey
Page 96: Adult root-boring weevil *Hylobius transversovittatus*; Bernd Blossey

2.17 Flowering Rush

Marc D. Bellaud: SOLitude Lake Management, Shrewsbury MA; mbellaud@solitudelake.com

Butomus umbellatus L; emergent shoreline plant in its own family, Butomaceae (flowering rush); originally placed in the Alismaceae (water-plantain) family
Derived from *bous* (Greek: ox) and *temno* (Greek: "I cut"), referring to its sword-like leaves with sharp edges that cut the mouths of cattle feeding on the species

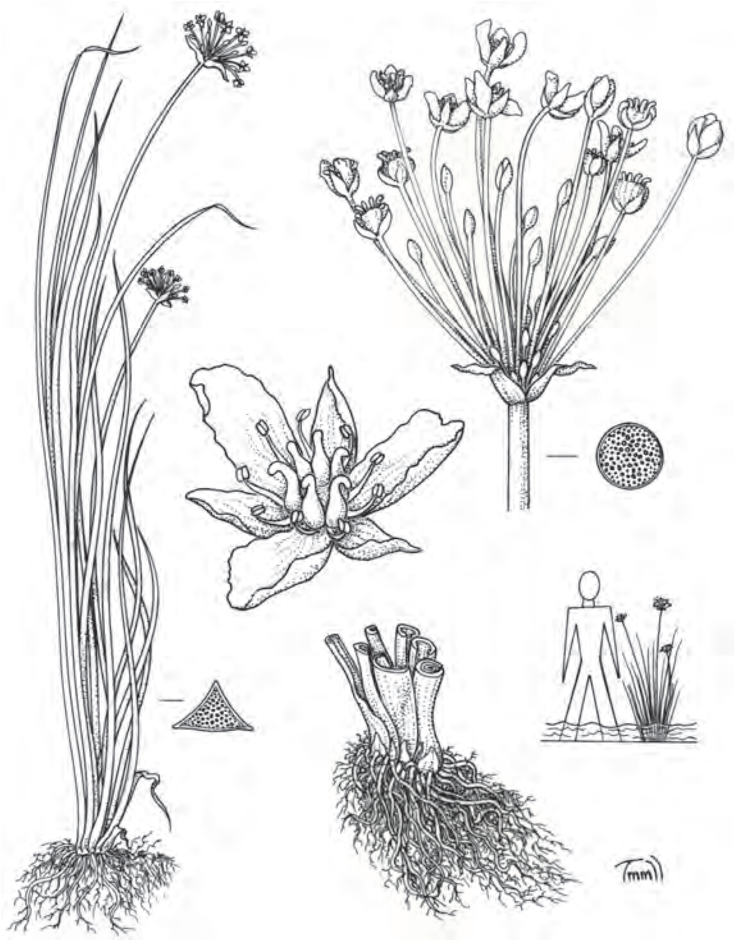
First identified along the St. Lawrence River in Quebec in 1897; likely introduced from Europe as a garden plant
Present in the northern US from Idaho to Maine and in the adjacent Canadian provinces

Introduction and spread

Flowering rush is native to Europe and Asia. It is thought that the species was first introduced to the US for use in ornamental gardens, but flowering rush thrives along shallow shorelines and in wetlands. The first observation of the species in North America occurred along the St. Lawrence River in Quebec in 1897 and botanists believe that multiple introductions have occurred since that time. By the mid-1950s, flowering rush populations were documented throughout the Great Lakes Region. Populations of flowering rush in the Great Lakes and points west are believed to be of European origin, whereas populations in the St. Lawrence River area are thought to be from Asia. Since the 1950s, flowering rush has spread to the west, north and east of the Great Lakes, with populations now found across the northern US and extending from Washington to Maine and nearly all of the adjacent Canadian provinces. Flowering rush tolerates a wide variety of shallow water and wetland settings and often forms dense stands that displace native riparian species, degrade fish and wildlife habitat, alter hydrologic patterns and interfere with recreational use of water bodies.

Description of the species

Flowering rush is a perennial monocot herb that can reach up to 5 feet in height. Plants have an extensive rhizome and root system and soil type or consistency and soil pH do not appear to affect growth. However, the species cannot grow in shade and is intolerant of saline or brackish waters. Plants become established in wet areas or along the shallow margins of lakes, ponds and streams and can grow into water up to 9 feet deep. Leaves of flowering rush are fleshy, thin and sword-like and resemble those of native bulrush (*Sparganium* spp.), but are triangular in cross-section. Submersed leaves remain limp or float on the surface of the water, whereas emergent leaves can reach to 3 feet in length and may have tips that are twisted in a spiral manner. Flowering rush is easiest to identify when it is flowering, which only occurs if plants are growing in very shallow water or along the shoreline. Plants flower between June and August, depending on temperature and latitude. The flowers are borne in an umbrella-shaped cluster (umbel). Individual flowers have three petals that are white to pink to purple in color.



Reproduction

Flowering rush is dispersed in four ways: seeds, vegetative bulblets produced on the inflorescence at the base of flower stalks, vegetative bulblets that form along the sides of rhizomes (underground stems with nodes that produce new shoots and roots), and rhizome fragments. Once established, the species expands its population size and spreads locally by rhizome elongation. Both seeds and bulblets can be transported by water currents and are long-lived, which facilitates their dispersal by wildlife, boaters and other human activities.

Eastern US populations of flowering rush are reportedly fertile diploids (with 2 sets of chromosomes), whereas sterile triploid populations (with 3 sets of chromosomes) occur in western North America. Diploid populations flower prolifically and produce both seeds and bulblets and their spread is due to dispersal of seeds and bulblets. Triploid populations in the West rarely flower and produce low numbers of seeds and bulblets. As a result, the majority of the spread of western populations is due to rhizome fragmentation, which results in clonal (genetically identical) populations.

Problems associated with flowering rush

Flowering rush can form dense infestations that compete with native riparian species and displace more desirable plants. Dense growth of the species may also allow it to outcompete threatened or endangered plant species and likely alters wildlife habitats. There are varying levels of concern about the impact of flowering rush on wetlands and fresh water habitats. For example, reports from the St. Lawrence River suggest that even high densities of flowering rush have not significantly reduced plant diversity. However, displacement of native plant species and the potential for wildlife habitat alteration make flowering rush a species of concern.

The impacts of flowering rush to water use and access may be more significant. For example, flowering rush has developed extensive monotypic populations in reservoirs with widely varying water levels in western states. The species is also currently causing economic impacts in irrigation canals and drainage ditches in the western US and large populations of flowering rush impede access to shallow lakes by colonizing shoreline areas where aquatic plants have not grown in the past. Marshlands are becoming dominated by flowering rush because the species thrives in areas with fluctuating water levels and expansion throughout littoral zones interferes with shoreline access, boating and fishing.

Management options

Active laboratory and field research on the management of flowering rush has been ongoing for the past several years, but information regarding the management of flowering rush infestations in North America is still sparse. However, the same management philosophies described for other species hold true – early detection of introductions and rapid response to new infestations provide the most effective control of flowering rush and limit further spread of the species. Flowering rush resembles many native species; therefore, accurate identification of the species is critical before initiating management efforts to avoid damaging nontarget desirable native plants. Flowering rush appears to thrive where bottom sediments are disturbed and following drawdown (Section 3.4), so preserving desirable aquatic plant populations is important to prevent establishment and spread of flowering rush.

Manual control methods include cutting (Section 3.5), hand digging and using bottom barriers (Section 3.4). These techniques are most appropriate for small infestations because rhizome fragments that are created can hasten spread. Bottom barrier can be used to control small areas of flowering rush through smothering, but this technique is not selective. Cutting will not kill flowering rush because the species will produce new growth from underground roots and rhizomes, but this method may decrease abundance and prevent seed and bulblet production by removing inflorescences. Plants should be cut below the water surface and care should be taken to remove all cut plant parts, especially rhizome fragments, from the water. Multiple cuts throughout the summer are required to provide adequate control and to prevent the formation of flowers, seeds and bulblets; also, cutting as frequently as every two weeks is necessary to reduce starch reserves in the roots and rhizomes. Large-scale cutting or harvesting efforts are often ineffective and may actually accelerate the spread of flowering rush due to the impacts to competing native plants, bottom disturbance and escaping plant fragments. Hand digging is useful only when managing individual plants or small infestations. The entire root structure must be carefully removed because fragments of roots, rhizomes or bulblets left in the sediment can rapidly regrow. All plant parts removed during cutting or hand digging must be taken out of the water and transported well away from water or wetland areas to prevent recolonization. There are currently no biological control agents (Section 3.6) that effectively control flowering rush, but research and exploration in the weed's native range of Europe and Asia is ongoing.

Herbicide treatment strategies (Section 3.7.1) include foliar applications, in-water applications and bare-ground (pre-emergent) applications during drawdown conditions. Foliar application of herbicides to control flowering rush is challenging; typically only a small part of the plant emerges above the water, so limited foliage is available for herbicide coverage and uptake, resulting in poor herbicide absorption and incomplete control. Several herbicides have been evaluated for foliar applications; of these, imazapyr has shown the greatest potential for control of above-water growth but successive years of treatment may be needed to reduce root and rhizome biomass. Diquat has been used for in-water treatments in midwestern and northwestern systems that are greater than one foot deep; one or two applications per summer control foliage and reduce root and rhizome biomass. Bare-ground treatments of imazapyr and imazamox applied during drawdowns are also effective, but two or more consecutive years of treatment may be needed to achieve significant root and rhizome reductions. There is no product that selectively controls flowering rush without the potential to harm other plants, so care must be taken during herbicide application to avoid impacts to nontarget species.

Summary

Flowering rush is an invasive species that has steadily expanded its range across the northern US and the Canadian provinces. It closely resembles bulrush and other native species and is difficult to identify unless it is flowering. The species employs multiple reproductive strategies that have helped to expand its range over the past 50 years. All potential impacts of this invasive species on aquatic systems are not yet known, but flowering rush is capable of abundant growth that can displace native species and alter habitats. Also, dense shoreline growth of the species can certainly interfere with access and recreational uses of infested water bodies. There is limited information available regarding the management of flowering rush, but as with other invasive species, early detection and rapid response are paramount to successfully controlling new infestations. Cutting below the water surface and careful hand-digging are effective on small infestations, while selective treatment with herbicides is currently the most effective strategy to control larger infestations of flowering rush. The expansion of flowering rush has occurred primarily in the western US and it is difficult to predict how extensive the problem may become, but research is ongoing to investigate the biology of the species and to identify additional management options.

Photo and illustration credits:

Page 97: Flowering rush; Thomas Woolf, Idaho State Department of Agriculture
Page 98: Line drawing; University of Florida Center for Aquatic and Invasive Plants
Page 99: Flowering rush; Thomas Woolf, Idaho State Department of Agriculture
Page 100: Flowering rush; Lyn Gettys, University of Florida

2.18 Ecology and Management of Algae and Harmful Algal Blooms

John H. Rodgers, Jr. (emeritus) and Megan Lee Pietruszewski: Clemson University, Clemson SC;
jrodgers@nctv.com, mpietru@clemson.edu

In contrast to the largely nonnative species of vascular aquatic plants in the preceding sections, most of the algal species featured in this section would be considered native to the United States or North America. Importantly, both native and non-native species of algae can develop nuisance-level populations and for some toxin producers, the density of algae that is problematic (dead fish or other animals) may not be discernable with unaided eyes.

Introduction

Algae (singular *alga*, Latin for "seaweed") are a large and diverse group of organisms that appear to be structurally simple, but range from small, unicellular species to large, multicellular forms, such as the giant kelps that grow to more than 200 feet in length. In fresh waters algae typically float in the water column (planktonic algae), form mats on the bottom of the waterbody (benthic or sediment associated algae) or form coatings on submersed structures (periphytic or attached algae). Relatively large freshwater macroalgae such as *Chara* and *Nitella* are often mistaken for vascular plants and can form rhizoids or root-like structures to anchor themselves in the sediments. Although the shapes and

sizes of algae range widely, they are considered structurally "simple" because their cells are not organized into the distinct organs such as roots, stems, leaves, flowers and fruits that are found in vascular plants. Algae are at the base of the food web and most are considered "primary producers" in aquatic systems because they provide sugars and chemical energy for other organisms (Section 1.1). Most algae are photosynthetic and use sunlight to "fix" carbon and produce sugars, but some unicellular species are unable to photosynthesize. Some algae live solely in fresh water while others live in salt water or marine systems. About 44,000 species of algae have been described by algal taxonomists, but more than 72,000 species of algae are thought to exist on earth, so many await discovery and description. While cyanobacteria (commonly called “blue-green algae”) have traditionally been considered algae, recent scientific studies usually exclude them due to important structural and physiological differences. However, for purposes of this discussion, cyanobacteria will be included as algae. In this section, approaches for intervention in harmful algal blooms (HABs), noxious growths of planktonic and filamentous cyanobacteria as well as some eukaryotic algae (species with distinct nuclei and chromosomes within a membrane) are reviewed.

Cyanobacteria or blue-green algae

Cyanobacteria are unicellular organisms that evolved billions of years ago and can be found in almost any aquatic habitat on the planet. Ancient blooms have been identified through sediment cores, suggesting that blue-green algae are

normal features of some lakes and have been for quite some time. Cyanobacteria perform three critical tasks: photosynthesis, respiration and nitrogen fixation. They can also control their buoyancy, allowing them to sink from the photic (upper) zone to the bottom of the lake and then back to the surface. Cyanobacteria can be particularly problematic because they may have a competitive advantage over most other phytoplankton. For example, some cyanobacteria can access pools of nitrogen and phosphorus that are not readily available to eukaryotic phytoplankton. Also, they can “hibernate” over winter and are rarely eaten by zooplankton, which allows populations to grow quickly. Blooms are not necessarily dependent on or caused by anthropogenic actions such as nutrient runoff but may be exacerbated by human activities. Limiting phosphorus inputs to water resources may reduce algal blooms in some cases but some cyanobacteria do not need high levels of phosphorus and nitrogen and can inhabit low-nutrient lakes.

Eukaryotes

Eukaryotic algae include green, red and brown algae, diatoms and dinoflagellates. Green algae, the basic components of the aquatic food chain, are commonly found in fresh water and may be inadvertently spread by humans; some – including *Cladophora*, *Codium*, *Caulerpa* and *Nitellopsis* – are invasive and can cause HABs. In contrast, most red and brown algae are marine. Some red algae are of commercial value and are harvested for pharmaceuticals and other products. For example, carrageenan (a common ingredient in lotions and food products) is from red algae and *Porphya* is a red alga used in sushi wrappers. Like green algae, most red algae are large, but some are coralline and provide important structural components in coral reefs. Brown algae include the “giant kelps”, which contain important pharmaceutical compounds and are commercially harvested for alginic acid. Diatoms are single-celled algae with “frustule”, or an outer cell wall made of silica, and about 20,000 species of diatoms that have been identified. Some diatoms are potent producers of secondary compounds, such as taste-and-odor compounds and toxins. Dinoflagellates have flagella (singular: flagellum), lash-like appendages that assist in propelling them through the water. They can grow in nutrient-poor waters and blooms are often stimulated without anthropogenic nutrient inputs. There are few reports of harmful freshwater dinoflagellates and the only species known to cause fish mortality is *Naiadinium polonicum* (formerly known as *Peridiniopsis polonica* or *P. polonicum*), a common and widespread species. Allelopathic effects (toxins produced by an alga that adversely affect another alga) by freshwater dinoflagellates are also rarely reported. Some dinoflagellates produce potent toxins at low population densities and do not discolor the water.

What causes algae blooms?

Algae are a critical part of a healthy aquatic ecosystem, but their normal presence and function in a water resource can cause problems when they grow out of control and form “blooms” or overgrowths of algae. Algae blooms have increased in recent decades and can occur due to increased sunlight, slow-moving waters and nutrient run-off. Eutrophication (the increase of nutrients in a water resource) is thought to be a leading contributor to bloom formation. Although eutrophication is a natural process, cultural eutrophication (the increase of nutrients to a lake from human causes such as agriculture and maintenance of lawns and landscapes) can also release phosphorus and nitrogen into watersheds and water resources. These excess nutrients can accumulate in lakes and reservoirs, resulting in increased frequency or intensity of algal blooms. Some noxious algae respond favorably to nutrients, while others (e.g., *Prymnesium*, *Didymosphenia geminata*) bloom as nutrient supplies are depleted.

Another factor that contributes to increasing algal blooms is the changing global climate. Climate change may affect algae growth because some species respond rapidly to changing conditions (warmer or colder temperatures, storms), which may contribute to their success in competing with other types of algae. For example, some species of cyanobacteria grow more rapidly in warmer temperatures. Noxious algae are also opportunistic and respond to episodic environmental events and upsets such as hurricanes and fires. In addition, we are moving algae around the planet at an unprecedented rate and transferring contaminated water (such as ship ballast water or from boat live wells) inadvertently spreads algal cells, fragments or spores. Finally, heightened awareness of algal blooms may also explain some of their apparent increase.

Problems associated with algae

Although algae occupy a critical niche in aquatic environments, many algae can rapidly grow to densities that become problematic or noxious. Noxious algal growths and HABs have compromised water resources throughout the world and have impeded the use of infested waters for wildlife, aquaculture, drinking, irrigation, recreation, navigation and industrial operations. Traditionally, HABs have involved high densities of algae and toxin production. However, density alone can be problematic even if toxins are not produced. Some algae may not achieve densities that are visible to casual observers and still produce sufficient toxins to kill most of the fish in the vicinity of a population growth or “bloom” (e.g., *Prymnesium* – see below). Other noxious algae can live on or adjacent to sediments and may not always be visible from the surface in waters that are not clear [e.g., *Lyngbya*, starry stonewort (Section 2.7), diatoms].

Excessive growths of algae change pH and water quality, reduce sunlight (which leads to low dissolved oxygen levels that can kill fish and other aquatic life), and cause foul tastes and odors. In addition, several groups of algae produce potent toxins that can be deadly in relatively small quantities. Left unmanaged, these algae can prevent the use of critical water resources for designated uses, thereby causing severe economic impacts. Annual losses of up to $2 billion in the US can arise from the inability to use a water resource for purposes such as domestic supply, industrial uses, irrigation, fire suppression and navigation, and can lead to declines in recreational uses and decreases in property values.

When do algae become problematic?

Although algae are critical components of aquatic ecosystems, their excessive growth allows them to outcompete native plants for sunlight, which can impair the habitat for fish and aquatic life. From a human perspective, algae maybe considered problematic when they become visible blooms or turn the water a pea-soup color. For example, Lake Okeechobee in Florida has received much attention for its green water. Some species of filamentous cyanobacteria may form mats along the surface of the water that are unattractive (as well as irritating for those unable to enjoy fishing, swimming or recreating due to limited mobility through the dense mats).

Many times, algae are considered problematic when population densities or production of toxins or other compounds interfere with the use of a water resource. Recreational activities such as fishing, boating and swimming can be impeded by algae forming mats along the surface of the water and dense stands below the water’s surface. Cyanobacteria produce a variety of taste and odor compounds that result in musty, sulfur, grassy or earthy odors in potable water but these are not known to be toxic. However, the negative effects of HABs go beyond aesthetics, taste and odor problems and pose severe risks to ecosystems and humans. For example, *Lyngbya wollei* is a mat-forming cyanobacterium that is native to North America but it is expanding its range from the southeastern US to the north, where it was recently detected in a lake near Detroit and in Lake Erie. Its thick mats prevent sunlight and oxygen from reaching other aquatic species, creating anoxic conditions and changing habitats and trophic networks. The increased occurrence of *Lyngbya wollei* has also been troubling for shoreline aesthetics. Rakes and other equipment have been used to remove *Lyngbya* mats from limited areas of shorelines. The loss of recreational activities has negative effects on property values and businesses (e.g., King’s Bay, Crystal River, Florida). Beyond unsightly mats, neurotoxins, hepatotoxins, and dermatotoxins have been identified in the mats. Toxins from *Lyngbya* have also been identified as tumor promoters, anti-fungal toxins, antiprotozoan toxins and mammal and fish toxins. Clearly, managing this alga when it blooms is important for human and ecosystem health!

Finally, and perhaps most importantly, HABs threaten human health. There are about 50 confirmed toxin-producing cyanobacteria and 100 known cyanotoxins, including neurotoxins, hepatotoxins, dermatotoxins, and endotoxin lipopolysaccharides (e.g., saxitoxins, anatoxin, cylindrospermopsins, lyngbyatoxins, hepatotoxins, and microcystins). One of these neurotoxins is beta-N-methyl-amino-L-alanine (BMAA), which has been linked to development of amyotrophic lateral sclerosis (ALS) and neurodegenerative disease. There are regions in New England where ALS cases are concentrated and higher than expected. Recent research using satellite remote sensing suggests that these ALS "hot spots" are concentrated in areas of poorer water quality, further suggesting linkages between water quality, cyanobacteria and high ALS incidence.

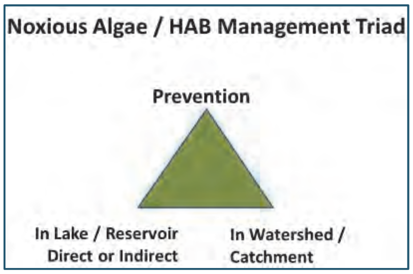
Animals as large as cattle and horses have died from exposures to cyanotoxins. Dogs swimming in water infested with cyanobacteria have died from anatoxin poisoning in California. In 2010, five dogs died and two people were hospitalized after swimming or water skiing in Milford Lake, Kansas. The only confirmed human deaths from exposure to cyanotoxins occurred in Caruaru, Brazil in 1996, when patients were inadvertently exposed to cyanobacterial toxin (microcystins) in the water used in dialysis treatment.

The effects of chronic cyanobacteria exposure on human health are largely unknown and the long-term effects are difficult to adequately study or quantify. Recent scientific studies suggest that exposure to cyanotoxins is not limited to direct exposure through activities like swimming, swallowing water or coming in contact with infested water, but people are also exposed to cyanobacteria through the air, known as aerosolization. Research is underway to understand exposures and health effects of cyanobacteria toxins and it is promising that these health conditions are receiving more attention from scientists and medical researchers.

Algae management

When noxious algae infest water resources, aquatic managers are faced with a decision to intervene or not. Unprepared, a manager may decide to take no immediate action but to monitor the situation and hope for better days. The decision to **not** intervene implies that the economic, human health and ecological risks and consequences due to algal blooms are acceptable. And "no decision" is a decision – ignoring the problem will not make it go away! It should be emphasized that taking no action is a decision and has consequences. For example, an unabated HAB will not wait on a decision and will continue to grow and spread. A few days or weeks of indecision can be very costly in terms of damage caused and increased costs for subsequent intervention or management. The time to plan is prior to arrival of the HAB.

There are a variety of options to manage algae but a decision to intervene usually requires a plan to be implemented in a timely fashion. It is important to note that the goal of modern algal management is not to remove all algae from impacted water resources, but to target the offending algae (called "targeted algal management"). As previously stated, algae are important in healthy aquatic ecosystems and some algal species are found in every water resource. Thus, a management goal is to restore uses of the water resource that are prohibited or limited by the noxious, harmful algae. Following a risk assessment, intervention involves altering the environment of the algae so the bloom is suppressed and the uses of the water resource are restored. An important point to emphasize initially is that algae are not uniformly distributed spatially or temporally. A lake may contain algae in certain locations and not in others, and at some times and not others. For example, some algae may be present at the surface of the lake. Other algae will hang out along the shoreline of the lake. Still other algae may live on one side of the lake, while the other side of the lake is relatively free from algae blooms. It is important to keep in mind that algae are not uniformly distributed as we discuss management strategies because different management strategies can be appropriate for different distributions of algae. In addition, if invasive algae have been detected in another water resource near your lake or water body, you can take measures to prevent the spread of algae to your lake.



Intervention should include both short-term and long-term solutions. Some short-term solutions may include tactics like algaecides, raking and aeration. The benefits of short-term solutions include seeing rapid results, which may include the extirpation of algae from the lake and restoration of water resources and uses. One common concern about implementing short-term solutions is that the underlying causes or "real" problem causing HABs, often presumed to be the buildup of phosphorus or other nutrients or climate change, is not addressed. Some options for intervention in growths of noxious algae are reviewed and contrasted below. When considering available tactics and developing a strategy that is specific for your situation, it can be helpful to be mindful of the HAB Management Triad.

There are two fundamental approaches for managing HABs: indirect and direct. Indirect approaches alter factors such as nutrients and light that can cause or promote algal growth, while direct approaches target specific problematic algae in the water resource or attempt to prevent the entry or spread of noxious algae into uninfested water resources. Indirect approaches involve tactics such as nutrient control in the water resource and watershed or catchment as well as management of littoral zones during construction of impoundments such as stormwater ponds and reservoirs. Indirect approaches generally do not focus on a particular species of algae or HAB. Direct approaches target specific problematic or recurring algae. No management option is a "silver bullet"; therefore, water resource and HAB management is necessarily adaptive and requires consideration of and use of all options as appropriate.

Resource managers recognize that algae must be managed in critical aquatic systems to maintain the designated uses of the water resource. When excessive algae growth occurs, adaptive water resource management is usually implemented to maintain the system and its uses. This involves careful consideration of all available options to manage or control algae and vascular aquatic plants to restore the uses of water resources. The unique characteristics of each water resource (including depth, latitude, altitude, shoreline, littoral zone, watershed, nutrients in sediments, etc.) also need to be considered in development of management strategy. Managing noxious algal growth requires actions that may include physical and cultural (Section 3.4), mechanical (Section 3.5), biological (Section 3.6) or chemical (Section 3.7.1) strategies alone or in combination.

Cultural and physical control

A popular notion is that algae can be controlled by controlling nutrient inputs to water resources. This idea perhaps originated from laboratory studies that showed that adding phosphorus and nitrogen resulted in proportional or incremental algal growth and was supported by addition of nutrients and observation of subsequent algal blooms in field studies. The time scale in these "experiments" was a few days to weeks (not centuries or thousands of years). The notion that followed was that removal of phosphorus from the water resource by decreasing inputs to the watershed or applying a binding agent such as alum or lanthanum to the water could control algal growth. And indeed that was the case in the simple laboratory studies using beakers or jars for testing. But aquatic systems store phosphorus in sediments and this phosphorus can be mobilized and made available for algal growth by winds, fires, turbulence, storm events, oxygen depletion in the hypolimnion and sediment feeding fish. This approach is not specific and targets algal production, so fisheries would also be generally impacted. Although watershed nutrient control is a popular notion and should be encouraged to slow cultural eutrophication, there is little scientific evidence that this is a successful approach for controlling noxious algal blooms.

This is not intended to discourage attempts to decrease nutrient loading to water resources, but outcomes of expensive and protracted efforts and expectations should be realistically considered. Unfortunately, preventing new phosphorus from reaching the water does not immediately reverse algal blooms because (as mentioned earlier) phosphorus is often stored in sediments. At best, nutrient reduction in inflows may initially slow algae growth; also, when developing management strategies for HABs in water resources, it is critical to consider not only management of nutrient loading from the watershed or catchment, but also how existing nutrients in the sediment of a water resource can be harnessed by cyanobacteria. Eutrophication emerges over time, so reducing inputs of nutrients into a lake will likely not reduce algae growth to a point where the uses of a water resource are restored until decades or hundreds of years have passed.

To summarize, long-term reduction of nutrient inputs to the water has been suggested as the “best” solution for harmful algal blooms and there is undoubtedly benefit to exploring the long-term causes and sources of HABs. In the meantime, there are concrete steps that can be taken immediately to address HABs and mitigate their harmful effects. These short-term solutions have benefits, even if they are not addressing the deeper causes of algal blooms, such as nutrient run-off from agricultural, suburban or urban areas. Individually or in combination, physical, mechanical, biological, chemical and preventative tactics can be used to reduce algal blooms and restore the water for recreational and other activities in a shorter time period than the long-term strategy of reducing nutrient inputs.

Light availability can have a significant effect on algae growth and new aquatic systems can be designed before construction to limit algal growth; for example, littoral zones can be limited by increasing water storage with deeper depths. These concepts are useful for stormwater impoundments but other factors such as access and recreation may take precedence over the algae and light consideration. Several dyes that have been registered by the United States Environmental Protection Agency (EPA) control algal growth by absorbing wavelengths of light that are needed for photosynthesis, but dyes are generally not used in flowing waters and larger aquatic ecosystems. The application of clays may flocculate and thereby remove algal cells from the water column may be a viable control tactic under some circumstances, but it is important to remember that clays and other flocculants are not registered as algaecides in the US. Also, rates of removal of cells due to clay flocculation, degree and rate of release of toxins from flocculated cells, physical and toxic effects on benthic organisms, and the consequences of organic loading from settled blooms on benthic oxygen conditions also need further evaluation. Benthic barriers have been used to control benthic HABs by blocking sunlight algae need to survive. These are challenging projects that require skilled divers and are used on relatively small areas such as around boat dock and marinas).

Raking or hand removal can be used to control small amounts of noxious algae growing in benthic areas or floating on the water’s surface, but this is a laborious process. Water manipulation (adjusting water flow and depth) is impractical for most systems and are only as reliable as the weather (if you need extreme drought or cold temperatures).

Biological control

There are very limited options available for biological control of algae despite their being impacted by bacteria, fungi, viruses and being eaten by zooplankton, fish, mollusks and snails. The addition of biological products to increase bacterial and fungal populations that compete with algae for available nutrients has been evaluated but the effectiveness of this strategy is not reliable or predictable. Aquatic habitat manipulation using tactics to increase zooplankton or other herbivore populations to reduce algae has long been studied. Larger herbivorous fish, crayfish, mollusks and snails consume algae in home aquaria but are subject to predation in natural systems; adding these aquatic animals is also site-specific, unpredictable and rarely feasible. Since each system is unique, efforts to use these types of bio-manipulation for algae control remain unpredictable and are the subject of considerable research.

Barley straw or straw from other cereal grains reportedly controls some algal species but results have been mixed. Some studies indicate that the straw releases anti-algal agents but these agents have not been positively identified; also, the addition of large amounts of organic matter to lakes and ponds has several deleterious effects. Any straw product that claims to control algae is violating the law administered by EPA regulations. This is not because straw is particularly dangerous, but rather because selling any product for algae, weed or insect control – even if that product is “natural” – when it is not registered as a pesticide with the EPA is illegal (Section 3.7).

Mechanical control

A variety of mechanical management tactics have been developed for HABs. The discussion in this section is limited to devices requiring external power (fossil fuel or solar) to function. These devices and approaches are not registered by the EPA and collateral damage associated with their use has not been thoroughly evaluated.

Harvesters range widely in size and design and are used mostly for filamentous algae such as *Lyngbya* and starry stonewort. Efficacy depends on the water resource, mobilization costs, access to the algae, access to remove harvested algae from the system, distance to disposal, design of the harvester and skill of the operator. Harvesters may be used in conjunction with algaecides to control areas not accessible to harvesting or to decrease biomass of algae prior to treatment. Rototilling or rotovating can be useful to manage HABs that grow as extensive mats on the sediment of lakes and reservoirs. Floating machinery can be used to "rototill" the sediments to depths of 7 to 9 inches, which dislodges algae and allows its collection and disposal. Similar to harvesting, this method does not discriminate between desirable and undesirable algae or plants and is limited to lakes and reservoirs with unobstructed areas and suitable sediments. Dragging chains attached to tractors on both sides of a canal is a common and effective technique for aquatic plant and benthic algae removal in canals if the canal banks are accessible. The dislodged algae and plant material are usually removed from the canal or the dislodged fragments will infest downstream areas. As with many control techniques, timing of the treatment influences efficacy. Like other harvesting operations, rapid regrowth necessitates repeat treatment. Dredging or excavating removes benthic algae from lakes, reservoirs and canals with a backhoe, dragline or similar excavating equipment. Significant drawbacks in using excavation equipment for algae control in canals include damage to the sediments and water resource profile and production of fragments or propagules and turbidity. Many noxious algae respond positively to disturbance like excavation, so rapid recolonization and regrowth means that ongoing management will likely be necessary and mechanical removal would have to be repeated frequently. The use of rototillers, chains, dredging or any other tactic that disturbs the sediments usually requires a Section 404 permit obtained from the Army Corps of Engineers as well as any permit required by the state.

Aeration may alter aqueous nutrient (phosphorus) concentrations or limit the time that algal cells spend in the upper photic zone on the water. By oxygenating the sediment-water interface, phosphorus release from sediments may be limited; also, aeration may disrupt the buoyancy and mobility of cyanobacteria. Scaling aeration to water resources of appropriate size and configuration is important to achieve success with this approach. Sonication or ultrasound can surely disrupt algal growth in the laboratory at a small scale, but implementation of this tactic in the field involves critical spatial and temporal scaling parameters. Sonication is advertised as effective, cost effective and environmentally sound for algal management. However, there have been reports of adverse impacts to nontarget species such as invertebrates (particularly zooplankton). The type and growth habit of algae appear to strongly affect efficacy. Considerable recent research has been ongoing regarding filtering or collecting algae for biomass and biofuel but isolating the algae from water has been a rate-limiting step. New technology involves dissolved gas floatation units, microstrainers, belt filters and settling ponds, while other approaches involve chemical coagulation and settling of algae. These approaches have not been successfully field-tested and operational data for filters and allied approaches for HAB management are not available.

Algaecides

Although it may be beneficial in the long run to reduce the human contributions (such as nutrient runoff) to algae blooms, many algal species can double their population size within two days or less, so immediate action is usually needed to manage infestations. In these time-sensitive situations, algaecides can serve as a first line of defense because they are cost effective, environmentally sound, socially accepted and work quickly to control excessive populations of algae. In order to efficiently and effectively use algaecides, water resource managers must rely on their knowledge of the aquatic system (i.e., nontarget species, water quality, etc.), the algae to be controlled and the algaecides labeled for use in their system.

Algaecides can be selective or non-selective against algae and range widely in their mechanisms of action. Selectivity depends on targeted algal species, location in the water resource, treatment and timing of application, product formulation and water chemistry. Algaecides must come in contact with algae (since there is little or no cell-to-cell movement of algaecides) and must enter algal cells to be effective. Algaecides differ in type of algae controlled, active ingredient, use sites, formulation, application rate, water use restrictions, dilution requirements and permit requirements. Algaecide active ingredients that are registered for use with the EPA include copper salts and formulations, organic compounds and peroxides (Section 3.7.1). Each algaecide has unique properties that should be carefully considered and evaluated prior to use in a water resource and some algaecides with the same active ingredients can differ in efficacy. Not all products are registered or available in all areas of the US. Always read and follow label instructions. The National Pollutant Discharge Elimination System (NPDES) requires a permit to apply algaecides and other pesticides over or near waters of the state or nation. For more information on which products are currently registered for control of algae in your state, check with your state regulatory authorities for product registration information and to determine which (if any) permits are required.

Algaecides are used primarily to control algal growth in impounded waters, lakes, ponds, reservoirs, stock tanks and irrigation conveyance systems. They can be applied as a spray directed onto an algal mat, sprayed or injected directly into the water column or applied as granular crystals or pellets. For successful treatments, it is important to get the algaecide to the targeted algae. Once a body of water becomes infested with algae, it is unlikely that algaecides will eliminate all algae or their spores (algae reproduce by cell division and/or by formation of spores). Due to their ability to reproduce quickly, however, algae are difficult to control in the long term and one treatment will rarely suffice. The efficacy of algaecides is short-lived in water and regrowth almost always occurs; as a result, re-treatment with algaecides is usually required.

One concern with any chemical control method is potential oxygen depletion after a treatment caused by the decomposition of the dead algae. Oxygen depletion can kill fish. If the water resource is heavily infested with algae it may be possible (depending on the algaecide chosen) to treat the algae in sections and let the algae in each section decompose for about two weeks before treating another section.

Applications of an algaecide can rapidly restore the uses of an aquatic system; adaptive water resource management should then be employed to develop strategies to prevent or mitigate future algal issues. Prevention measures such as the control of algal movement in bilge waters of boats and bait buckets could be undertaken. Other practices, such as reduction or elimination of runoff and nutrient control in the watershed, may be helpful in the long term, but are unlikely to provide near-term relief for excessive algae problems.

Algal toxins in freshwater systems

This section will focus on some toxin-producing species of freshwater algae as well as other noxious algae which can adversely affect other algae, vascular plants, invertebrates, fish and mammals. Algal toxins are problematic in fresh waters when they are produced in sufficient quantities with sufficient potency to cause direct toxicity to organisms, decrease feeding and growth rates and cause food safety issues. Production of algal toxins may be associated with a "bloom" or exceptionally dense growth or accumulation of algae. The term "harmful algal bloom" (HAB) has been used to describe a proliferation, or "bloom", usually of phytoplankton. Because phytoplankton serves as the base of most aquatic food webs, the impact of these blooms can be devastating for consumers throughout the food web and for other flora and fauna in the affected ecosystem. Even severe blooms of non-toxic algal species can spell disaster for animals in freshwater aquatic systems since massive quantities of phytoplankton can deplete oxygen in shallow waters.

The species of freshwater algae that cause HABs, as well as their effects, vary widely. While some are toxic only when they achieve high densities, others can be toxic at very low densities (only a few cells per liter). Whereas some blooms discolor the water (thus the terms "green scum", "red tide" and "brown tide"), others are almost undetectable by unaided visual observation. The effects of HABs generally fall into two major categories: 1) public health and ecosystem effects, and 2) economic impacts. Broadly, public health and ecosystem effects can include factors such as:

1. Filter feeding shellfish (e.g. clams, mussels) may accumulate algal toxins by feeding on the toxic phytoplankton, sometimes to levels potentially lethal to humans or other consumers;
2. Potential fish, shellfish and bird kills, occasionally invertebrate and mammal kills;
3. Decreased light penetration can alter ecosystem function and structure;
4. Discoloration of water can be aesthetically unpleasant;
5. Toxins or other compounds released by the algae can kill fauna directly or result in low oxygen conditions as the bloom biomass decays (especially critical where fauna cannot escape the area);

6. Blooms can be harmful to other algae or primary producers and the food webs that are dependent on them; and
7. The effects on shoreline residents of long-term or chronic exposures to algal toxins.

Direct economic impacts caused by HABs include loss of income for commercial fishermen, loss of food for subsistence fishermen and consumer concerns regarding food safety, as well as declines in property values. Some examples of management of algae producing toxins and noxious algae in freshwater systems in the US would perhaps be useful. The chapter is limited to toxins produced by cyanobacteria, golden algae and euglenoids. Other algae (e.g., *Chrysochromulina* and others) that produce both toxins and/or taste-and-odor compounds can be important, but are not included in this discussion. Also, some more recent discoveries, such as the *Stigonematales*-like cyanobacterium (*Aetokthonos hydrillicola*) that has been implicated in avian vacuolar myelinopathy, are not included since sufficient information for management has not been developed at this time.

Cyanobacteria: the blue-green algae

Cyanobacteria (blue-green algae) are geologically ancient, broadly distributed inhabitants of fresh, brackish, marine and hypersaline waters, as well as terrestrial environments, and grow in diverse habitats ranging from thermal springs to the

arctic. Although cyanobacteria are classified as bacteria as opposed to algae, they are photosynthetic in aquatic systems. In fact, cyanobacteria are much larger than other bacteria and are major contributors to global photosynthesis and nitrogen fixation. Cyanobacteria occur in unicellular, colonial and filamentous forms; they grow in a wide variety of conditions and can rapidly become the dominant algae in nutrient-rich water bodies. Cyanobacteria can form blooms so thick that the surface of the water appears to be covered with blue-green paint. Several cyanobacteria in the US produce substances that cause taste and odor problems in water supplies and aquaculture. Some blue-green algae, particularly *Anabaena, Planktothrix* and *Microcystis*, are widely distributed in the US and can produce toxins that are poisonous to fish and wildlife that drink toxin-contaminated water. There are documented cases of blue-green algal toxins harming humans that have consumed or inhaled toxin-tainted waters.

Cyanobacterial ecology in freshwater systems

Cyanobacteria are most abundant in eutrophic conditions, but they can readily colonize most freshwater systems and can rapidly grow to readily visible masses or “blooms” that render the water resource unstable, unreliable or unusable. The occurrence and abundance of particular cyanobacteria in a freshwater system depend on a variety of ecological factors, including nutrient status, salinity, light conditions, turbulence and mixing, temperature and herbivory. In some freshwater systems, true algae may grow faster than cyanobacteria. However, cyanobacteria can seize the advantage in eutrophic situations by out-competing algae for nutrients, thriving in low dissolved oxygen and photosynthesizing more efficiently at lower light levels. Cyanobacteria are also less affected by turbidity, high concentrations of ammonia and warmer temperatures than are algae; in addition, they may produce chemicals (toxins) that inhibit the growth of competing algae and reduce grazing by invertebrates.

Cyanobacterial toxins in freshwater systems

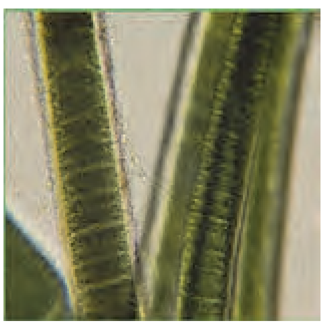
Several types of cyanobacterial toxins are produced by various species of blue-green algae, but most cyanotoxins are classified as either neurotoxins or hepatotoxins. Neurotoxins attack the nervous systems of vertebrates and invertebrates; symptoms of neurotoxin poisoning in fish include loss of coordination, twitching, irregular gill movement, tremors, altered swimming and convulsions before death by respiratory arrest. Neurotoxins are produced by several genera of cyanobacteria including *Anabaena, Aphanizomenon, Microcystis, Planktothrix, Raphidiopsis, Arthrospira, Cylindrospermum, Phormidium* and *Oscillatoria*. Neurotoxins produced by *Anabaena* spp., *Oscillatoria* spp. and *Aphanizomenon flos-aquae* are responsible for animal poisonings around the world. Hepatotoxins ultimately lead to liver failure; symptoms in fish include flared gills (due to difficulty breathing) and weakness or inability to swim, which can result in mortality within 24 hours of exposure. Cyanobacterial hepatotoxins are produced by many genera of cyanobacteria, including *Microcystis, Anabaena, Planktothrix, Nostoc, Oscillatoria, Anabaenopsis, Dolichospermum, Aphanizomenon, Phormidium* and *Cylindrospermopsis*. Hepatotoxins such as microcystins, have been implicated in deaths of fish, birds, wild animals, and agricultural livestock, and are responsible for human illness and deaths in India, China, Australia and Brazil.

Management of toxic cyanobacteria

Toxin production does not always occur in a bloom of toxin-producing cyanobacteria but toxins can quickly be produced in toxic amounts by high-density blooms of cyanobacteria. The decision to treat cyanobacteria with an algaecide is prompted by a variety of factors, including the size of the affected water resource, the number and type of organisms (e.g., fish, mammals) in the system, the age and condition of the organisms that will be potentially affected, the sensitivity of the target cyanobacterium to treatment and the cost of treatment. Most toxin-producing cyanobacteria are susceptible to algaecide treatments but some experimentation may be needed to identify the best treatment for a specific strain at a site. Occasionally, the idea that algal cells may leak toxins is proposed as a consideration for initiating – or choosing not to initiate – an algaecide treatment, but the idea that all algaecides cause toxin leakage in all situations is not supported by existing data. Also, algae can double their population densities in two to three days, and toxin production is often proportional to density, so choosing not to treat suggests that the risks associated with further production of toxin are acceptable. There is no way that treatment can increase the concentration of total toxin; however, failure to treat toxin-producing algae can result in increased exposure to toxins and associated risks. Management techniques other than algaecides may be considered as well. Tactics that have been tried include physical mixing and aeration of water, increasing flow rate or flushing to decrease hydraulic retention time, and decreasing or altering nutrient content and composition. Some of these options are site-dependent and therefore may or may not be viable, depending upon the site and situation.

Lyngbya wollei

Lyngbya wollei (lyngbya) is a filamentous, mat-forming cyanobacterium that occurs in a variety of fresh waters. Based on modern genetic techniques, *L. wollei* has recently been assigned a new name (*Microseira wollei*) that may be accepted with time. It can grow in waters with low nitrogen concentrations due to its ability to fix atmospheric nitrogen,



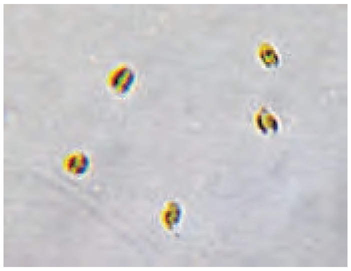
thrives at extreme temperatures ranging from melt-water lakes and streams to hot springs and contains photosynthetic accessory pigments (i.e., phycobilins) that permit growth in extremely low light conditions. Unlike other algae, lyngbya persists throughout the year and infestations are becoming more common throughout the southeastern United States. For example, it is a nuisance in the Everglades as well as Rainbow and Crystal Rivers in Florida, Guntersville Reservoir in Alabama and the lower Rio Grande, Texas. Lyngbya filaments are usually not branched and are covered by a polysaccharide sheath. Lyngbya forms dark blue to black benthic mats that range in thickness from several inches to several feet thick and may cover small ponds and entire coves. These mats, which are composed of entangled filaments and can achieve dry weights of up to 4.5 tons per acre, can trap gasses and float to the water's surface where they impede navigation and recreation, cover and smother submersed plants and clog water intakes. In addition, lyngbya releases a strong and unpleasant earthy or musk-like odor. Lyngbya also produces several toxins including paralytic shellfish poisons. If benthic algae such as lyngbya interfere with critical water resource usages and problems become severe, water resource managers often are compelled to intervene with management techniques.

Several approaches have been used to manage excessive growths of lyngbya, including chemical (algaecides), harvesting and benthic barriers. Control efforts for this species have often been hampered by the thick sheath of mucilage that surrounds the algal cells and the presence of both surface and benthic mats in the spring and summer. Based on economic and environmental considerations, copper-based algaecide formulations are often used. For copper-containing algaecides, water characteristics and other site parameters influence the speciation of copper and thus the bioavailability and efficacy of an algaecide application. Some control success has been achieved using copper-based algaecides combined with penetrants.

Prymnesiophytes: the golden-brown algae

Most toxin-producing species in the genus *Pymnesium* form harmful blooms in brackish water, but strains are expanding into freshwaters, especially during droughts. *Prymnesium parvum* is a relatively small (~10 microns), saltwater-loving organism that is commonly referred to as "golden algae." Golden algae are widely distributed and blooms in brackish and inland waters have been responsible for mass die-offs of fish and significant economic losses in Europe, North

America and other continents. The species is capable of photosynthesis but also feeds on bacteria and microorganisms. Dense growths of golden algae may color the water yellow to copper-brown or rust and the water may foam if aerated or agitated. *Prymnesium* has spread to several freshwater systems in the US, possibly due to drought conditions.



Golden algae produces at least three toxins which alter cell membrane permeability and are collectively known as prymnesins. The toxins produced by *Prymnesium* cause fish to behave erratically and young fish are more sensitive than their elders. Affected fish may have blood in gills, fins and scales and they may be covered with mucus. Fish may move to the shallows of tainted waters and leap from the water in an attempt to escape exposure to the toxins. Gill repair can occur within hours if fish are moved to uncontaminated water during the early stages of exposure but moving affected fish to other systems may also spread golden algae to previously uninfected systems. Mammals and birds often eat dead fish and drink water in the area but aquatic insects, birds and mammals are reportedly not affected by prymnesin toxins. The golden alga is not known to harm humans, but dead or dying fish should not be used for human consumption as a precautionary measure.

Texas has been impacted by recurrent golden algae blooms in several reservoirs and rivers and Texas Parks and Wildlife has offered some detailed advice regarding management options (Sager et al. 2007), but the reader is cautioned that some methods used in aquaculture and private pond settings may be illegal elsewhere. Algaecides that have been used to manage golden algae in isolated pond culture include ammonium sulfate and copper sulfate; however, the concentration of ammonium sulfate required to control *P. parvum* (~0.17 mg/L of unionized ammonia) may adversely affect some fish and copper sulfate may kill desirable algae along with golden algae, thus decreasing food resources for zooplankton and disrupting fish feeding. In Chinese aquaculture of carp, suspended solids (mud), organic fertilizer (manure) and decreased salinity have been used to control *P. parvum* (although these are not EPA registered algaecides), with the best results from decreased salinity and ammonium sulfate. In addition, Rodgers et al. (2010) found that *Prymnesium* from several locations was controlled by 200 ug/L of chelated copper.

Euglenoids

Euglena is a genus of widely distributed algae found in many shallow, relatively calm, eutrophic freshwater systems throughout the US. Species of *Euglena* are sources of ichthyotoxin (a suspected neurotoxin) in freshwater aquaculture and have caused mortalities in striped bass, channel catfish, tilapia and sheepshead minnows. For example, a number of outbreaks of toxic *E. sanguinea* have occurred since 1991 in hybrid striped bass production ponds in North Carolina and have resulted in the loss of more than 20,000 pounds of fish due to complete kill in affected ponds. Symptoms of exposure to *Euglena* toxins begin with the fish going off its feed for no apparent reason. Within 24 hours of cessation of feeding, gills become reddened, fish swim at or near the surface in an agitated or disorientated state (often with the dorsal fin extending out of the water), swim on their sides or even swim upside down. If steps are not taken immediately after observing this state, the fish will be dead within 24 hours.

If a toxic *Euglena* bloom is suspected, the pond should not be aerated because this will disperse the bloom throughout the pond. Species of *Euglena* are exceptionally mobile and as the toxicity event progresses to the point where exposed fish are disorientated, the highest concentration of toxins seems to occur in the downwind side of the pond. Euglenoids are sensitive to several commercially available algaecides, particularly those with labels that specify that euglenoid algae are susceptible. In the past, species of *Euglena* have responded to treatments with chelated copper formulations at 0.12 to 0.5 mg/L, as well as to peroxide formulations at or below the maximum label rate.

Best management practices for noxious algae

As adaptive water resource management is practiced today, adhering to best management practices for noxious algae involves the following:

1. <u>Accurate diagnosis of the problem in a water resource</u> requires representative samples of water or benthic material containing the potential noxious alga(e).

2. Identification of the targeted alga(e) and distribution by microscopic confirmation of the density or toxin or taste-and-odor compound production. Algae are not usually uniformly distributed in aquatic systems; they may be "layered" in the water column, mixed by the wind or may be in benthic patches.
3. Measurement of water characteristics for the site can influence algal growth as well as compatibility and performance of a treatment option (e.g., algaecide). The minimum data set needed typically includes temperature, pH, hardness, conductivity and alkalinity. Other information such as nutrient concentrations and suspended solids may be useful as well.
4. Site characteristics are important for discerning an appropriate and compatible approach based on water depth and area, as well as the designated uses for the water resource (e.g., drinking water supply, swimming, fishing, etc.). Site history such as previous use of algaecides and the frequency and intensity of noxious algal blooms would be useful.
5. Evaluation of potential options should be considered in terms of their compatibility with the site and situation, as well as their ability to achieve the desired outcomes. For example, a dye to block sunlight may be appropriate for a fountain or contained water body where the entire system can be treated, but may not very useful or efficient in systems where considerable water exchange occurs. As another example, NSF-certified algaecides may be required for drinking water resources.
6. Selection of an option or options may require some experimentation to select an appropriate option. Responses of target algae to algaecide exposures can differ due to formulation or application technique.
7. Application of the selected option to achieve the required exposure (often called dose, treatment or rate), which is crucial to the success of a treatment [achieving the desired response from the target alga(e)]. The goal is to treat the target alga(e), not necessarily the water.
8. Monitoring results is an important step in adaptive water resource management that provides information to guide future decisions.

Summary

Algae are a large, diverse group of organisms that range from small, unicellular species to large, multicellular forms. They may float in the water column, form mats on the bottom of the waterbody, form coatings on submersed structures or appear similar to vascular plants. Algae form the base of the food web and most are considered primary producers in aquatic systems, so they play an important role in the ecosystem. However, the overgrowth of algae can result in blooms that can be noxious and may interfere with ecosystem services and human uses of the affected water. Some species of algae produce toxins that can be harmful to fish, wildlife and humans and must be managed to reduce or prevent these problems. There are a number of tactics that can be employed for algae management but the use of algaecides is typically the most effective and economical method to control noxious and harmful algae blooms. As more water resources are impacted by noxious algae and as these resources are increasingly utilized for critical purposes such as drinking water supply, irrigation and habitat for fish and wildlife, management of these crucial freshwater resources will become more prevalent. The need to constantly innovate and improve our approaches is clear and that is the goal of adaptive water resource management and BMPs.

NOTE: If an algaecide application is indicated, all regulatory approvals and permits must be obtained. Following label instructions and restrictions is necessary to comply with federal law. Mention of a control tactic for toxin-producing algae does not constitute endorsement of an algaecide or any other tactic for your specific situation. Check with your local extension agent regarding site-specific permit requirements and restrictions.

Photo and illustration credits:

Page 101: *Euglena sanguinea* bloom on a pond in SC; John Rodgers, Clemson University
Page 102: *Microcystis aeruginosa* along the shoreline of Pawnee Lake, NE; John Rodgers, Clemson University
Page 104: Floating mats of *Lyngbya wollei* at Kings Bay/Crystal River, FL; John Rodgers, Clemson University
Page 105: HAB Management Triad; John Rodgers, Clemson University
Page 107: Aerator; William Haller, University of Florida
Page 109: *Planktothrix*; John Rodgers, Clemson University
Page 110: *Lyngbya wollei*; John Rodgers, Clemson University
Page 111 upper: Photomicrograph of *Prymnesium parvum* from Dunkard Creek, WV; John Rodgers, Clemson University
Page 111 lower: Photomicrograph of *Euglena sanguinea* from a pond in SC; John Rodgers, Clemson University

3.1 A Manager's Definition of Aquatic Plant Control

Jeffrey Schardt: Florida Fish and Wildlife Conservation Commission (retired), Thomasville GA; jeff.schardt@gmail.com
Michael D. Netherland: US Army Engineer Research and Development Center (deceased)

Introduction

It would seem like a simple task to define "control", but this section illustrates how difficult and variable the term can be. Even scientists argue about the definition of control. For example, entomologists who release a potential biocontrol agent (Section 3.6) may define control as a 10% reduction in plant growth, but most lake managers and homeowners would disagree. Do barley straw, enzymes and bacteria really control algae (Section 2.18)? Can native insect populations be augmented to provide weed control? Much depends on the definition of "control".

Defining aquatic plant control

During the past few decades demand for access to and use of US surface waters has increased. These uses include real estate, recreation, irrigation, hydropower, potable water, navigation and efforts to conserve environmental attributes

such as fish and wildlife habitat. Aquatic plants are a natural and important component of many freshwater systems and resource managers consider a diverse assemblage and a moderate level of aquatic vegetation to be beneficial for numerous ecosystem functions. Nonetheless, an overabundance of aquatic plants, particularly invasive nonnative plants, can impair freshwater systems, requiring some level of aquatic plant management to conserve water body uses and functions. These aquatic plant management activities routinely take place on water bodies ranging in size from small private ponds to large public multi-purpose lakes and reservoirs.

With increasing demands and values associated with surface waters has come a greater need for aquatic plant control. Nonetheless, the term "control" can take on many meanings depending upon the type and amount of use of each water body, the species of plants present, the responsibilities of resource managers and the objectives of various stakeholder groups associated with the water body. A quick review of reference materials provides the reader with dozens of descriptions and synonyms for "control", and yet for various reasons none provide a meaningful definition for aquatic plant management. The Aquatic Plant Management Society (APMS) has requested that we address this deficiency by providing an aquatic plant manager's working definition of aquatic plant control.

While the terms aquatic plant control and aquatic plant management are often considered synonymous, many resource managers consider control efforts as being operational in nature and management as a process more aligned with program goals and objectives. The APMS defines aquatic plant control as "*techniques used alone or in combination that result in a timely, consistent and substantial reduction of a target plant population to levels that alleviate an existing or potential impairment to the uses or functions of the water body*".

This definition best applies to management techniques that directly target a reduction in plant biomass. It is recognized that some management strategies seek to impact factors such as plant reproductive capacity (e.g., production of flowers, seeds, tubers, etc.) or nutrient availability. While these techniques are often recognized as a valuable component of an integrated management program, physical reduction of plant biomass may not result for many years. Moreover, in our definition, the use of the term "substantial" may seem ambiguous; however, we feel there is an inherent problem with using quantitative guidelines (e.g., a 70% reduction in biomass results in acceptable control) to define what is in most cases a series of qualitative field observations by the aquatic resource manager and stakeholders to determine the success of the management activity. Aquatic resource managers should always consider if the proposed management technique has a successful track record and know the limitations of the potential strategy. Claims that a product or technique can provide control should be supported by peer-reviewed literature, experiences from other resource managers with similar management objectives or current research and demonstration efforts.

No single definition of aquatic plant control can cover each specific contingency; therefore, good communication on the front end is key. Resource managers and stakeholders must first establish expectations for the amount and duration of plant control prior to the initiation of a control activity and then implement a management strategy to meet these expectations. This definition and the following discussion are intended to address factors that relate directly and indirectly to aquatic plant control. Numerous variables influence aquatic plant control operations and many of these parameters, including water body uses, environmental conditions and available management tools, are presented throughout this handbook, along with the influences they may have on the planning or outcomes of aquatic plant control operations. This information may be useful to managers responsible for conserving identified uses and functions of public waterways and who must explain to stakeholders the reasoning behind management plan selection and the ultimate results.

Linking management decisions to aquatic plant control expectations: factors that influence decisions and outcomes

Aquatic plants have been controlled in US surface freshwaters under organized programs for more than a century, so it is natural to ask why it is necessary at this point to provide a definition of aquatic plant control. In questioning a number of managers, researchers and other stakeholders, it became obvious that opinions on what constituted acceptable control of an aquatic plant population varied widely. While agricultural managers have been using terms such as "weed free periods" and "crop yield reductions" to define the economic benefits of weed control in cropping systems, aquatic plant managers have a different focus than their terrestrial counterparts. Agricultural weed managers usually attempt to control a broad spectrum of weeds to enhance one or more crop species in a fairly controlled environment with a specific function. Aquatic plant managers usually try to control one or two weeds (usually invasive exotic species) to conserve or enhance perhaps dozens of desirable plants as well as multiple uses of aquatic systems. In essence, an agricultural definition of "weed control" does not encompass the issues associated with aquatic plant management.

In developing a manager's definition of control, it was initially tempting to utilize the language of research to provide a quantitative definition. Both the amount and duration of plant control can be readily quantified within the framework of an experimental study or demonstration project. Nonetheless, many experimental studies result in destructive sampling of the target plants at a given point in time (e.g., 90% reduction 8 weeks after treatment) and they often don't allow us to determine if even better control or subsequent recovery would result at a later point in time. While this efficacy information can be very useful to managers regarding the expected performance of a management technique, the uses, functions and environmental conditions can vary widely among water bodies and within water bodies through time. This will influence not only the level of management that may be attempted, but also the outcomes of each control operation. While research projects utilize methods that allow for quantification of control, the vast majority of aquatic plant control operations are ultimately judged by fairly subjective visual observations and qualitative means (e.g., the target plants are near the bottom, difficult to find and the current level of control is rated as good). Therefore, plant control or lack thereof is largely based on whether or not resource manager and stakeholder expectations have been met.

As noted above, there are numerous issues that either directly or indirectly influence aquatic plant control and management strategies. Before selecting control tools or developing management strategies, three key elements should be addressed that will ultimately influence the manager's decision making process.

Native vs. nonnative vs. invasive aquatic plant control

The National Invasive Species Council defines an invasive species as an alien species whose introduction does or is likely to cause economic or environmental harm or harm to human health. While there are major distinctions between invasive exotic and native species, the main objective of this paper is to clarify the term "control" and as such will not make significant distinctions between managing invasive exotic species and nuisance growth of native plants. Whether a plant is a native or exotic, it can cause problems for given water uses (e.g., water conveyance, access, recreation). Nevertheless, two key distinctions between nuisance native and invasive plants deserve further discussion. First, problems associated with nuisance native vegetation are typically site-specific, whereas invasive plants can impair uses and functions of waters across a broad spectrum of conditions and on a regional scale. The vast majority of large-scale aquatic plant control efforts in the US target invasive species. These plants have the potential to spread and dominate new ecosystems and they also have demonstrated the ability to become established in relatively stable aquatic systems. The philosophy behind invasive plant management programs often is to reduce the potential for spread within and among water bodies by reducing the plant biomass to the greatest extent practicable. The second distinction involves early detection and rapid response (EDRR) programs. These efforts are typically unique to invasive exotic species. A significant and costly multi-agency effort may be initiated to control a very small infestation; however, given the potential negative properties of many invasive exotic plant species, these front-end efforts are viewed as necessary and cost-effective.

Efficacy vs. control

It is tempting to define aquatic plant control in terms of an expected percent reduction in coverage or biomass of a target plant population. Some regulatory agencies (e.g., California EPA, Canada Pest Management Regulatory Agency) require that herbicide manufacturers prove the efficacy of their products prior to registration. In this regulatory scenario, a product must reduce a target pest population by greater than 70 or 80% to provide efficacy. A farmer, for example, expects control or weed suppression to levels that allow maximum crop yield over a 3 or 4 month period from planting to harvest. Within the discipline of aquatic plant management, numerous techniques can provide both a rapid and significant reduction in a target plant population (> 70%), but these results may only be sustained for a few weeks or months. Therefore, depending upon when the efficacy of a management technique is measured, one assessment may suggest that control was achieved, whereas a subsequent assessment conducted weeks, months or a season later may lead to the conclusion that the management effort failed to provide an acceptable level of control.

If resource managers and stakeholders have agreed to implement a strategy to provide an entire season of biomass reduction and the target plants recover within one or two months, then by our definition, control has not been achieved. In contrast, some methods may result in slow initial impact on a weed population but may ultimately provide one or more seasons of control. To complicate matters, many stakeholders fail to grasp that an aquatic plant problem may require more than one treatment or strategy. It is the responsibility of resource managers to understand the strengths and weaknesses of the various management techniques and then convey this information to the stakeholders. If expectations are not defined properly, stakeholders may lose confidence in the management program. When managers do not establish clear expectations, they are often questioned as to whether control was achieved. Attempting to assess aquatic plant control when clear expectations were not established on the front end is one of the biggest challenges in coming up with a meaningful definition or even assessment of control.

Environmental controls

Managers must be careful not to confuse slow-acting control methods with natural variations in plant populations. While it is often tempting to link a prior control effort with the large-scale decline of a target plant population, environmental events (e.g., droughts, floods, hurricanes, seasonal senescence, etc.) often are largely responsible for these declines. If sufficient data do not exist to support a cause and effect relationship between a control effort and plant biomass decline, managers should avoid making claims that cannot be supported by evidence. Some managers rely on environmental events (e.g., flooding events that scour submersed plants or move floating vegetation; prolonged periods of high, dark water that prevent light penetration for submersed plants) to provide control. While this can be effective, in order to be considered an aquatic plant management technique, there should be some level of predictability associated with the environmental event. From a management perspective there is a big difference in relying on routine seasonal flooding events to control a given plant population versus relying on 100-year floods or droughts to provide plant control.

Levels of aquatic plant control

At the most basic level, there are three possible aquatic plant control approaches:

1) no attempt to control
2) control efforts to eradicate a plant species
3) some level of intermediate control that is either incomplete or temporary

No attempt to control

Despite its connotation, the "no control" option is a valid management decision whose potential outcomes must be considered by managers and explained to stakeholders. Factors that influence a manager not taking active control measures may include:

- plant species – Is the plant invasive? Is it a native plant impairing water body uses or is it just unwanted by stakeholders?
- size of infestation – Is this a pioneer infestation consisting of a few plants? Is it an established, but stable, population? Is it an established population or starting to approach problematic thresholds?
- plant location – Is the infestation in an isolated location? Is the location conducive to spreading the pest plant by fragmentation, flow, etc.? Are there important nearby water bodies that are prone to becoming infested?
- plant biology – Is there a likelihood of a rapid population expansion? Would "no control" permit the plant to produce viable seed or vegetative propagules that could make later control efforts more difficult and expensive?
- exploitation – Is the plant species providing an ecological service (e.g., nutrient uptake, food source for waterfowl, habitat for fisheries, etc.)?
- managerial will – Managers may be under pressure to not control a plant because it provides benefits (perceived or real) to a user group. Stakeholders may oppose control because they are not familiar with proposed methods or do not understand an invasive plant's growth potential.
- managerial experience – Inexperienced resource managers are often uncomfortable with making aquatic plant management decisions (especially on a large scale). Until a manager understands the issues and situation, the "no control" option may be viewed as the safest and least controversial.

The consideration of these factors and others may justify a "no control" decision. There are consequences associated with all management decisions and "no control" is not exempt. As previously addressed, plant reductions related to environmental factors could be included within the realm of the "no control" option. While environmental events such as floods, droughts, freezes or severe algae blooms can be quite effective in reducing aquatic plant biomass, these events are not typically predictable and they are not initiated by managers. Nonetheless, the fact that some managers tend to rely on seasonal or weather events to provide effective control suggests the term "no control" may be a misnomer in these situations.

Eradication

Much like defining control, eradication has proven to have numerous meanings among managers, researchers and stakeholders. In a strict sense, eradication means the complete and permanent removal of all viable propagules of a plant population. This is confounded when a population is removed and then reintroduced at a later time. Some plants may be eradicated following single management efforts [e.g., removal of waterhyacinth (Section 2.11) plants prior to seed set], whereas others such as hydrilla (Section 2.2) or giant salvinia (Section 2.13) may requires years of intense surveillance and management. Eradication efforts are typically employed when a region, state or watershed is threatened with a new introduction of an invasive species that has potential for significant economic or environmental impact. Based on efforts by various resource management agencies to date, aquatic plant eradication programs are characterized by:

- sustained and multi-year efforts to insure elimination of the plant population
- small-scale efforts to control relatively few plants
- control costs on a per acre basis can be quite high
- the overall impact of repeated control efforts on the infested water body is continually weighed against the regional threat posed by the invasive plant
- control efforts may eventually be reduced; however, vigilant monitoring remains a key to success

Temporary control

Outside the realm of eradication, all other control efforts are temporary. Temporary control is essentially an acknowledgement that 100% control is either not an economically viable management objective or is not possible. Temporary control is a continuum that can be represented by the short-term reduction of target plants following mechanical harvesting or spot treatments with contact herbicides to many years of control that may result from grass carp (Section 3.6.2) stocking for submersed plants or decades of suppression of alligatorweed (*Alternanthera philoxeroides*) by the alligatorweed flea beetle (Section 3.6.1). Thus, temporary control results when the aquatic plant manager has made the decision that eradication is not a viable endpoint and some level of target plant persistence is acceptable in the management strategy for a given water body.

Temporary control is achievable using a variety of methods. Managers should evaluate each proposed method and the integration of various methods in terms of meeting specific control objectives.

Maintenance control

Maintenance control is applied on a lake-wide or regional scale over time, usually to reduce and contain invasive species. Once established, invasive aquatic plants can be extremely difficult, if not impossible, to eradicate. However, managing invasive plants at some prescribed level that does not impair the uses and functions of the water body can reduce environmental and economic impacts. As the term implies, maintenance control indicates that a conscious decision has been made to actively control an aquatic plant problem with the understanding that a long-term commitment to management rather than eradication is the goal. Simply stated, maintenance control involves routine, recurring control efforts to suppress a problem aquatic plant population at an acceptable level.

Maintenance control encompasses a continuum of control objectives. On one extreme, the goal of maintenance control may be to reduce and sustain a plant population at the lowest feasible level that technology, finances and conditions will allow. This strategy has proven effective in managing established populations of highly invasive aquatic plants. By managing waterhyacinth at low levels through frequent small-scale control operations, there is a corresponding reduction in the overall management effort, especially herbicide use and management costs. There also are environmental gains, such as reductions in sedimentation and dissolved oxygen depressions and minimal impacts to existing habitat. At the other end of the spectrum, maintenance control operations can be applied just prior to plant populations impairing the uses or functions of the water body. This strategy entails allowing plants to grow to the brink of problem levels and therefore may be best employed to control slow-growing or otherwise non-invasive plants.

Paradoxically, there is often more stakeholder support for crisis management (allowing plants to reach some problem or impairment level) than maintaining invasive species at low levels. This may be related to stakeholders being unaware of invasive plant growth potential. It also may be related to the public’s perceptions of control methods – for example, not understanding that less herbicide (and less funds) may be needed to maintain plants at low levels rather than waiting for an obvious problem to develop.

Adaptive management

Since maintenance control represents a long-term commitment, it must also encompass a strategy known as adaptive management. Uses and functions of water bodies change through time, as do conditions within water bodies and among plant populations. Examples include target and nontarget plant growth stages, water temperature, depth, clarity and flow. All change several times during the year and can require different control strategies or different expectations for control outcomes. Therefore, integrated management plans for each aquatic plant control operation must account for and adapt to these changes.

Communicating control expectations to user groups

Many stakeholders view aquatic plant management endeavors as a one-time control effort with no further need for additional management. This does not reflect the reality of the discipline of aquatic plant management. The vast majority of management programs require a sustained effort over multiple years to keep unwanted vegetation under control. For example, while grass carp can provide long-term control of hydrilla, this result is due to their continuous presence and feeding on any plant regrowth. Carp can sustain control for many years, yet removal of the carp due to natural losses or on purpose will typically result in the recovery of the target plant. Likewise, a single treatment with the herbicide fluridone (Section 3.7.1) may remove a target invasive plant such as Eurasian watermilfoil (Section 2.3) within a system for one to several years. Upon discovery of new plants, many stakeholders are dismayed that the treatment did not eradicate the problem. In some cases, these plants may have regrown from seed or they may have been introduced from a nearby lake or reservoir that was not managed. Aside from the use of an effective classical biological control organism (highly selective – Section 3.6) or high stocking rates of grass carp (non-selective – Section 3.6.2), user groups must be informed about the importance of maintaining continuity in an aquatic plant management program. Single small-scale efforts that don't address the problem at an adequate scale often lead to claims that "we tried that, and it didn't work." A lake full of hydrilla or Eurasian watermilfoil may require whole-lake management efforts. The control may last one, two or more seasons, but experience suggests that these invasive plants will ultimately return.

Photo and illustration credits:

Page 113: Diverse aquatic plant community; Lyn Gettys, University of Florida

3.2 Developing a Lake Management Plan

John D. Madsen: USDA Agricultural Research Service, Davis CA; jmadsen@ucdavis.edu

Introduction

Invasive aquatic plants are a major problem for the management of water resources in the United States. Nonnative invasive species cause most of the nuisance problems in larger waterways and often produce widespread dense beds that obstruct navigation, recreation, fishing and swimming and interfere with hydropower generation. In addition, dense nuisance plants increase the likelihood of flooding and aid in the spread of insect-borne diseases. Invasive plants also reduce both water quality and property values for shoreline owners.

Invasive species have a negative impact on the ecological properties of the water resource. They may degrade water quality and reduce species diversity while suppressing the growth of desirable native plants. Invasive species may alter the predator/prey relationship between game fish and their forage base, which results in higher populations of small game fish. Invasive species may also change ecosystem services of water resources by altering nutrient cycling patterns and sedimentation rates and by increasing internal loading of nutrients.

The most troublesome invasive plants that cause problems in the United States and recommendations for managing them are discussed in Section 2 of this manual. These exotic weeds are most likely to cause the greatest concerns, but many other native and nonnative species can cause problems as well, particularly in small areas or in ponds.

Development of a management plan

Water resource managers need to have an aquatic plant management plan for long-term management, even in bodies of water that have not yet been invaded by these exotic species. An effective aquatic plant management plan should establish protocols to prevent the introduction of nuisance plants, provide an early detection and rapid response program for the waterbody so new introductions can be managed quickly at minimal cost and aid in identifying problems at an early stage. The plan should also assist in identifying resources and stakeholders so that coalitions can be built to aid in the management of problem species. The planning process should include information that is already available and identify gaps in knowledge where more information is needed. An effective management plan will help water resource managers communicate the need for management of invasive species and provide a rationale or approach for management. A comprehensive aquatic plant management plan should have eight components: prevention, problem assessment, project management, monitoring, education, management goals, site-specific management and evaluation.

Prevention

The focus of a prevention program is education and quarantine combined with proactive management of new infestations [early detection and rapid response (EDRR)]. Most invasive aquatic plants are introduced to a water body as a result of human activity and introductions most often occur when invasive plants are transported on boats, watercraft and boat trailers. Prevention activities can include signage at boat launches and marinas and other educational programs. Successful prevention programs utilize federal and state legislation, enforcement, educational programs in broadcast and print media and volunteer monitoring programs. An early detection and rapid response program should be employed in conjunction with prevention efforts to control new infestations at an early stage. Proactively controlling new infestations before they develop into large populations of exotic plants is both technically easier and less expensive, which results in major cost savings in the long run. The eradication of small populations is much more likely than eradication of large established populations. Early detection and rapid response is a critical component of an exotic species prevention program and is emphasized by federal agencies involved in invasive species management.

Problem assessment

Problem assessment should focus on identifying a problem in a given waterbody and collecting information about the problem. This information can then be used to formulate specific problem statements that define the cause of the problem. Problem assessment is the process of both acquiring objective information about the problem, such as maps and data on plant distribution, and identifying groups or stakeholders that should have input into formulating the problem statement. Problem assessments should also identify the causes of the problem and should increase the understanding of the water resource by reviewing information that is already available and highlighting areas where additional information is needed. A specific problem statement should be developed using the resources identified during problem assessment to aid in refining the concerns of users and the nature of the nuisance problem.

Project management

Project management is often a neglected aspect of managing invasive plants, particularly when volunteers manage the project. Successful projects are the result of good planning and management of assets, which include financial resources, partnerships, volunteers and other personnel. Detailed records of expenses must be maintained, particularly if the project is funded by government entities. In addition, a thorough evaluation of success of the program should include expenditures of both time and labor.

Monitoring

A monitoring program should include not only an assessment of the distribution of the target plant species, but also a program to monitor other biological communities (including desirable native plant communities) in the water body. Water quality parameters should be recorded on a regular basis to determine whether long-term changes have taken place in the water body and to assess whether management activities have had a positive or negative effect on other aspects of the water resource. Monitoring should also include baseline data collection (as outlined in the problem assessment section above), compliance monitoring involving a permit and assessments of management impacts to the environment at large. Successful monitoring programs often include a "citizen" monitoring component. For instance, citizen monitors have assessed water quality in many water bodies for several decades using techniques as simple as measuring water clarity using a Secchi disk (see page 2). The largest volunteer network in the US is The Secchi Dip-In (https://www.nalms.org/secchidipin/), though many states also have a statewide volunteer network (e.g., Florida LakeWatch; http://lakewatch.ifas.ufl.edu).

Education and outreach

Education and outreach should be initiated at the beginning of the program and should continue throughout the project. Education initially consists of familiarizing the project group with the problem and possible solutions, which helps to build a consensus regarding the solution. As the program progresses, education efforts should be extended to include the public (in addition to stakeholders in the lake association) and to inform them of the problem, possible solutions and what actions the program is taking to address the problem. It is important to provide as much information as possible to the public and to be forthright and open about management activities. A public web page devoted to the management program can be a very successful tool but the project group should utilize local media outlets, such as newspapers and radio, as well. Also, if your project is successful, share your success with others through homeowners associations, state environmental agencies or your local county cooperative extension service.

Plant information and methods

The development of a program to monitor invasive plants requires a list of invasive, nonnative, native, endangered and threatened plant species in the waterbody, maps marked with the locations of species of concern or species targeted for management, locations of nuisance growth and bathymetric maps. Quantitative plant data (sampling for plant distribution or abundance using a recognized sampling protocol) should be used for assessment, monitoring and evaluation as often as possible. Quantitative data are more desirable than qualitative data (subjective assessments such as "a big population" or "heavily infested") because:

- Quantitative data are objective and provide hard evidence regarding the distribution and abundance of plants, whereas subjective surveys are based on opinion rather than fact
- Quantitative data allow for rigorous statistical evaluation of plant trends in assessment, monitoring and evaluation
- Quantitative data and surveys may eliminate costly but ineffective techniques in a given management approach
- Quantitative data allow individuals other than the observer to evaluate the data and to develop their own conclusions based on assessment, monitoring and evaluation data

Plant quantification techniques vary in their purpose, scale and intensity (see table below). Cover techniques include both point and line intercept methods. These techniques yield the most information regarding species diversity and distribution and can reveal small changes in plant community composition. The best method for measuring plant abundance remains biomass measurement but this is time-intensive and usually reserved to evaluate the effectiveness of management activities. Hydroacoustic surveys measure submersed plant canopies while the plants are still underwater and are excellent for assessing the underwater distribution and abundance of submersed plants; however, this technique is unable to discriminate among species. Visual remote sensing techniques, whether from aircraft or satellite, have also been widely used to map topped-out submersed plants or floating and emergent plants.

Aquatic Plant Quantification Techniques

Technique	Information produced
Cover techniques: point intercept	Species composition and distribution (whole-lake)
Cover techniques: line intercept	Species composition and distribution (study plot)
Abundance techniques: biomass	Species composition and abundance
Hydroacoustic techniques: SAVEWS	Distribution and abundance (no discrimination among species)
Remote sensing: satellite, aircraft	Distribution (plants near the surface only, no discrimination among species)

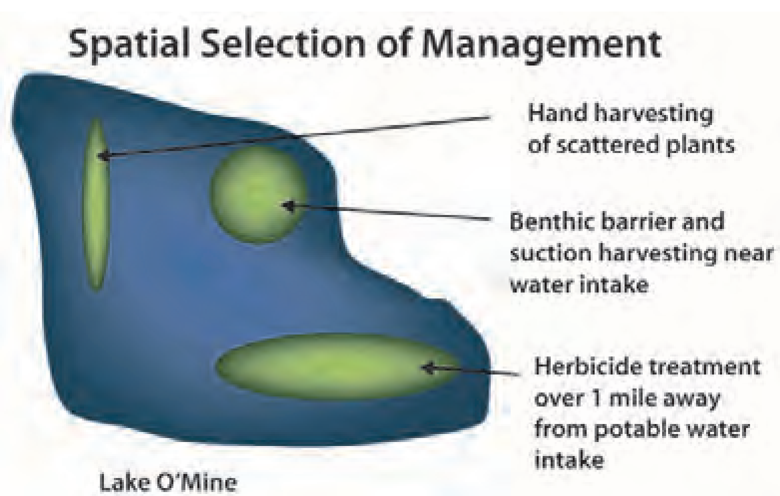
Management goals

Specific management goals that are reasonable and testable should be formulated as part of the management plan. This set of goals provides the milestones that can be used to determine whether the management program is successful. If specific management goals are not established, stakeholders may dispute whether management efforts have been successful since they may lack a clear understanding of the expectations of the management program (Section 3.1). Goals should be as specific as possible, including indicating areas that have a higher management priority.

Providing stakeholders with a specific set of goals will allow them to evaluate quantitative data to determine whether management goals have been met. For instance, if vegetation obstructs recreational use of the waterbody, a goal of "unobstructed navigation" is vague and may result in unending management. If, however, the goal is to maintain navigation channels in navigable condition 90% of the time, then the success of the management program can be measured, tested and compared to the specific goal. Once plant management goals are developed, methods to achieve the goals should be implemented using techniques that are acceptable to stakeholders and regulatory agencies based on environmental, economic and efficiency standards. Management techniques will vary based on conditions within the water body and frequently change over time; this is referred to as site-specific management.

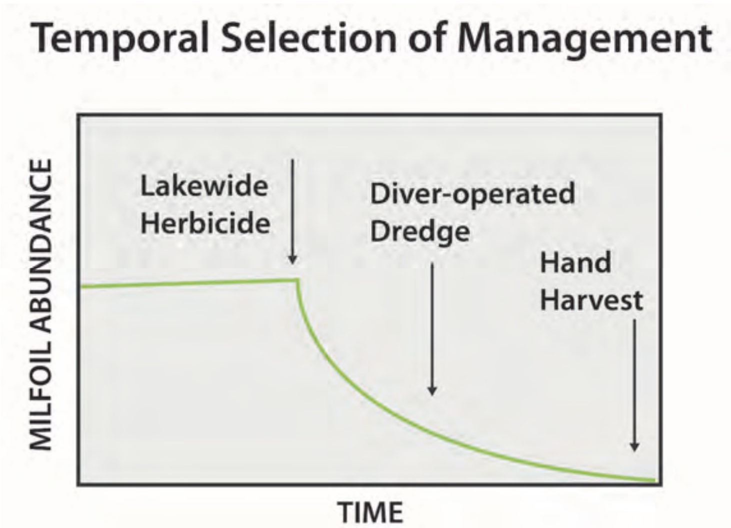
Site-specific management

Site-specific management utilizes management techniques that are selected based on their technical merits and are suited to the needs of a particular location at a particular point in time. Techniques should be selected based on the priority of the site, environmental and regulatory constraints of the site and the potential of the technique to control plants under the site's particular conditions.



Spatial selection criteria include the identity of the target weed species, the density of the weed, the size of the infested area, water flow characteristics, other uses of the area and potential conflicts between water use and restrictions associated with selected management techniques. For example, consider an area of nuisance growth that is close to a drinking or irrigation water intake. The primary use of the water (i.e., drinking or irrigation) may preclude the use of herbicides that cannot be applied to waters used for drinking or irrigation; therefore, the most appropriate control method for this area might be the use of a benthic barrier and suction harvesting. Consider another site that is more than a mile from the same intake. Weeds at this site could be controlled with herbicides without restrictions on other uses (provided the label specifies use of the herbicide in the area). Perhaps you have an area that is colonized mainly by scattered plants instead of dense stands. If the goal is to eradicate the plant from the water body and you have volunteers at your disposal, hand pulling may be the best method to prevent the formation of dense beds of the weeds.

Management techniques may change over time based on the success (or failure) of the management program. For example, consider Long Lake in Washington State, a small body of water that was dominated by Eurasian watermilfoil (Section 2.3) throughout more than 90% of the littoral zone. A whole-lake treatment of fluridone was applied to Long Lake, which reduced the biomass of the weeds by more than 90%. Small remaining beds in the second year were managed with diver-operated suction harvesting, benthic barriers or spot treatment with contact herbicides. By the third or fourth year, routine surveys found only sporadic Eurasian watermilfoil fragments, which were removed by hand harvesting. Similar treatment programs have been successful in other water bodies as well, which demonstrates that it is appropriate to alter management techniques as weed control requirements change over time. A wide variety of aquatic plant management techniques may be employed and include physical (Section 3.4), mechanical (Section 3.5), biological (Section 3.6) and chemical (Section 3.7.1) control methods. Regardless of method, all techniques should be selected based on their technical merits, as limited by economic and environmental thresholds.



Evaluation

Evaluation of management techniques and programs is typically lacking, even in large-scale management programs. A quantitative assessment should be made to determine the effectiveness of weed management activities, identify environmental impacts (both positive and negative) of management activities, provide the economic cost per acre of management and address stakeholder satisfaction.

Summary

It is critically important to develop a management plan to effectively prevent and control invasive aquatic plants in water resources. Planning should be a continuous process that is ongoing and evolves based on past successes and failures. A comprehensive plan should educate the public about invasive species so they can identify and exclude weeds from uninfested areas. Aquatic plant management programs should also provide a concise assessment of the problem, outline methods and techniques that will be employed to control the weed and clearly define the goals of the program. Mechanisms for monitoring and evaluation should be developed as well and information gathered during these efforts should be used to implement site-specific management and to optimize management efforts. The planning process helps to prepare for the unexpected in weed management, but resource managers should expect the plan to change as stakeholders provide input and management activities commence.

Photo and illustration credits:
Page 121: Nuisance growth near a water intake; John Madsen, USDA ARS, Davis, CA
Page 122: Long Lake herbicide treatment; John Madsen, USDA ARS, Davis, CA

3.3 The Endangered Species Act

Patricia Shaw-Allen: NOAA Fisheries Office of Protected Resources, Washington DC; pat.shaw-allen@noaa.gov

Introduction

The goal of the Endangered Species Act (ESA) is to protect and recover species that are endangered or threatened to the point where protection under the ESA is no longer necessary. This requires the removal or reduction of threats to the species and the ecosystems on which they depend. Control of native and nonnative nuisance aquatic vegetation, even when the intent is to improve aquatic habitat, can result in harm to ESA-listed species and trigger the need for careful planning and permitting under the ESA. This section first provides an overview of how species and habitats are protected under the ESA, then discusses how to pursue aquatic vegetation control activities in compliance with the ESA.

Listing species as endangered or threatened species and designating critical habitat

The United States Fish and Wildlife Service (USFWS) and National Oceanic and Atmospheric Administration's National Marine Fisheries Service (NOAA Fisheries) (taken together: the Services) share responsibility for implementing the ESA. The Services identify those species that need protection, or “listing”, under the ESA based on the best available scientific and commercial data. Species are listed through a rulemaking process that invites public input. The Services encourage protection of candidate species or species of concern through proactive conservation programs to educate the public, stimulate research to fill information gaps and foster voluntary efforts to conserve species before they require listing.

Generally, the USFWS is responsible for terrestrial and freshwater species while NOAA Fisheries is responsible for most marine species and fish species that use both marine and freshwaters (e.g., Pacific salmonids, Atlantic sturgeon). The USFWS does have jurisdiction for certain marine mammal species, including walrus, sea otters, manatees and polar bears. The Services also share responsibility for certain species such as sea turtles, which nest on land, and some fish species that use both marine and freshwater habitats (Atlantic salmon and Gulf sturgeon, for example). At the time of this writing, approximately 1,600 species that occur within the US (including US territories and the Commonwealth of the Northern Marianas) are listed as endangered or threatened under the ESA.

The ESA requires that listing determinations be based solely on the best scientific and commercial information available. Other factors, including economic impacts, cannot be considered when making species listing determinations under the ESA. Critical habitat is designated, to the maximum extent prudent and determinable, concurrently with the species’ listing. Unlike the listing determination, economic impacts, national security impacts and any other relevant impacts are considered when designating critical habitat.

Section 9 of the ESA prohibits the “take” of listed species and Section 4(d) allows for protective regulations, including the application of Section 9 take prohibitions for threatened species. “Harm” is defined by a 1999 modification to the regulations by both Services as any act which actually kills or injures fish or wildlife, and emphasizes that such acts may include significant habitat modification or degradation that significantly impairs essential behavioral patterns of fish or wildlife. Take resulting from a pesticide application includes potentially adverse impacts to a listed species in the area where physical, chemical and biological changes may occur as a result of that application, not just the immediate footprint of the application.

Complying with the Endangered Species Act

The ESA Section 9 prohibition on take applies to all individuals, organizations and agencies subject to United States jurisdiction. However, there are two mechanisms in the ESA where take may be exempted or authorized. A Section 10 incidental take permit may be sought by a non-federal entity for take of ESA-protected species resulting incidental to, but not for the purpose of, carrying out the covered activity. Permits are not required for activities that are authorized, funded or undertaken by a federal agency when take has been evaluated through Section 7 consultation and described in the Incidental Take Statement of a Biological Opinion (see below).

Staff at the Services field and regional offices will provide technical assistance in determining whether an aquatic plant control effort may result in take of ESA-protected species and identify ways to prevent or minimize take so that a permit may be granted. If take of some USFWS and some NOAA Fisheries species may occur, a permit will be required from each agency to cover take of species under their respective jurisdictions. The types of permits a non-federal entity may apply for include:

Permits for scientific research or enhancement of the survival of an ESA-listed species. Permits issued under Section 10(a)(1)(A) of the ESA are for activities resulting in intentional take that involve research designed to increase our knowledge of an ESA-listed species or activities that increase survival through reducing threats, improving habitat or a breeding program. Enhancement of survival permits are issued by the Services to non-federal landowners participating in Safe Harbor Agreements or Candidate Conservation Agreements with Assurances. These agreements encourage landowners to take actions to benefit species while also providing assurances that they will not be subject to additional regulatory restrictions as a result of their conservation actions. Section 10(a)(1)(A) permits issued by the USFWS also include recovery and interstate commerce permits. Recovery permits allow for take as part of activities intended to foster the recovery of listed species, including research to better understand the species' long-term survival needs, whereas interstate commerce permits issued by the USFWS allow transport and sale of listed species across state lines for purposes such as a breeding program.

Incidental take permits are issued by the Services under Section 10(a)(1)(B) of the ESA. If a non-federal entity believes their otherwise lawful activities may result in take of endangered or threatened species, they may choose to apply for an incidental take permit and develop a Habitat Conservation Plan (HCP). An applicant is required to develop a HCP as part of their incidental take permit application to demonstrate how they will meet the permit issuance criteria. For instance, the HCP typically describes the covered activity, impacts to the species, minimization and mitigation measures, funding assurances and plans for monitoring the effectiveness of the HCP.

If aquatic vegetation control activities will occur in waters occupied by ESA-listed species and no other federal permits are required, consult the appropriate NMFS or USFWS field office for assistance and guidance before choosing to seek an incidental take permit. Other permits may be appropriate for efforts that are intended to restore or improve habitat used by ESA-listed species.

Permits for scientific research or enhancement of survival

Enhancement of survival permits are provided to non-federal landowners who have entered into Safe Harbor or Candidate Conservation Agreements with Assurances. In a Safe Harbor Agreement, the landowner agrees to maintain, create, restore or improve habitat for endangered or threatened species. Working with the landowner, the Services will establish a baseline condition for each species and determine whether the proposed actions will result in a net conservation benefit. The landowner may then incidentally take listed species, as long as baseline conditions are maintained. Taking below the baseline is sometimes permitted if the taking does not have a significant adverse effect on the baseline and is likely to provide a long-term benefit on the baseline.

A Candidate Conservation Agreement with Assurances is a formal agreement between the Services and one or more parties to address the conservation needs of proposed or candidate species (or species likely to become candidates) before they become listed as endangered or threatened. Landowners voluntarily commit to conservation actions that will help stabilize or restore the species with the goal that listing will become unnecessary. If the conservation actions are not sufficient to prevent ESA-listing of the species, the Agreement automatically becomes a permit authorizing the landowner incidental take of the species. Thus, the agreements provide landowners with assurances that their conservation efforts will not result in future regulatory obligations in excess of those they adopt at the time they enter into the Agreement. The Agreement also provides an avenue to potential federal or state cost-share programs for those landowners who want to conserve the species or want to manage habitat on their land.

Incidental take permits

To avoid potential violation of Section 9 of the ESA, a non-federal entity may voluntarily apply for an incidental take permit in cases where their actions may result in take that is incidental to, and not the purpose of, an otherwise lawful activity. It is up to the party responsible for a planned activity to decide whether to seek an incidental take permit. If an incidental take permit is not obtained and take occurs, the responsible party is liable for that take under the enforcement provisions of the ESA.

Before deciding to pursue an incidental take permit, a prospective applicant should request assistance from the appropriate NMFS or USFWS field office because there are many options for applicants to consider. Once they decide to seek a permit, the applicant should allow adequate time to complete an application and develop the HCP. It is important to fill out the entire application to provide a complete description of the planned activity. For example, if a section of the permit form is not applicable to a proposed activity, the applicant will need to explain why the section is not applicable on the form. Once an application is submitted, the Services may request additional information. The Services cannot process an application until all required information is provided.

Once an application for an incidental take permit has been accepted, it will be posted on-line and a notice soliciting public comment within a 30-day period is published in the Federal Register. The Services review comments submitted by the public and provide the permit applicant the opportunity to respond to substantive comments. The decision to issue or deny a permit is based on the permit application, public and expert comments, the permit applicant's responses to those comments and the environmental analyses of the requested activities. Once a permit is issued, it is valid until the specified expiration date. All permits require annual reports on incidental take and some permits require additional reporting.

Incidental take permit applications must include a conservation plan or an HCP. HCPs can provide for partnerships between the Services and non-federal parties to conserve the ecosystems upon which listed species depend, ultimately contributing to their recovery. An HCP is designed to offset the harmful effects a proposed activity might have on listed species and provide additional conservation benefits. It also provides flexibility for the landowner by including planning for unlisted species and species that are proposed for listing under the ESA. This proactive approach advances the goals of the ESA by possibly preventing the need to list a species. It also can be effective in moderating future conflicts by fostering strong partnerships.

In order to obtain a permit, the applicant's application and HCP must meet the following permit issuance criteria by demonstrating that:

i) the taking will be incidental;
ii) the applicant will, to the maximum extent practicable, minimize and mitigate the impacts of the taking;
iii) the applicant will ensure that adequate funding for the plan will be provided;
iv) the taking will not appreciably reduce the likelihood of the survival and recovery of the species in the wild; and
v) other necessary measures will be met.

To accomplish the above, an HCP must include a sufficient description of the anticipated impact of the proposed activity on ESA-listed species and its habitat. This includes:

- the estimated number of animals of the listed species and, if applicable, the subspecies or population group and range;
- the type of anticipated taking, such as harassment, predation, competition for space and food, etc.;
- the effects of the take on the listed species, such as descaling, altered spawning activities, potential for mortality, etc.;
- the anticipated impact of the proposed activity on the habitat of the species; and
- the likelihood of restoration of the affected habitat.

The HCP needs to identify the steps that will be taken to monitor, minimize and mitigate such impacts, including detailed monitoring plans, specialized equipment, methods of conducting activities or other means along with the funding available to implement measures taken to monitor, minimize and mitigate impacts. The HCP also needs to identify the alternative actions that would result in less take or no take that were considered and the reasons why those alternatives are not being used. A list of all sources of data and personal communications with recognized experts used in preparation of the plan must be included with the HCP.

Working with federal agencies

Under Section 7 of the ESA, any federal agency proposing to authorize, fund or carry out an action that may affect ESA-listed species is required to consult with the Services. Since the outcome of Section 7 consultations can affect the activities of non-federal entities working for or with a federal agency or seeking authorization or funding from a federal agency, the federal agency is responsible for determining the status of that entity as an “Applicant” under Section 3(13) of the ESA. As an Applicant, the non-federal participant associated with the federal action has the opportunity to submit information for consideration in the consultation, review and comment on biological opinion drafts, contribute to the development of Reasonable and Prudent Alternatives (if needed) and reject extensions of the consultation period greater than 60 days.

During consultation, the Services determine whether the proposed action is likely to jeopardize the continued existence of ESA-protected species or adversely modify designated critical habitat. If the activity is likely to jeopardize a species or adversely modify designated critical habitat, the Services prescribe Reasonable and Prudent Alternatives to the proposed action to prevent jeopardy and adverse modification or destruction of designated critical habitat. If the action is likely to adversely affect, but not jeopardize species or adversely modify or destroy designated critical habitat, the Services prescribe Reasonable and Prudent Measures to reduce take incidental to the action. These prescriptions are mandatory if the action is to proceed in compliance with the ESA. The analyses that support these requirements are documented in Biological Opinions issued in completion of Section 7 consultation.

Summary

The control of aquatic plants in waters where ESA-protected species and habitats occur may require permitting under Section 10 of the ESA or an exemption from the take prohibitions through an Incidental Take Statement issued pursuant to an ESA Section 7 consultation between the Services and a federal agency. Technical assistance from the Services will help determine whether take of ESA-listed resources may occur and identify ways to minimize or avoid take. It is up to the party responsible for a control activity to obtain the necessary permits or exemptions when take is anticipated. Incidental take permits issued to non-federal entities can cover take that occurs during aquatic plant control activities that are lawful, but not conducted to conserve ESA-protected resources. Enhancement of survival permits provide for safe harbor and candidate conservation agreements with assurances protecting the rights of landowners who are pursuing aquatic plant control activities intended to advance conservation of ESA-listed species. When working for or with a federal agency, take coverage is specified in the Incidental Take Statement of a Biological Opinion documenting an ESA Section 7 consultation between the federal agency and the Services.

For more information

More information on the ESA and ESA-protected species and habitats is available online at:

https://www.fws.gov/endangered/index.html and
https://www.fisheries.noaa.gov/topic/endangered-speciesconservation

While these sites include resources that help determine whether ESA-protected species are in your planned pesticide application area, it is always a good idea to check with local USFWS or NOAA Fisheries biologists during the planning phase for any aquatic plant control activity.

Are USFWS ESA-listed species or designated critical habitat present?

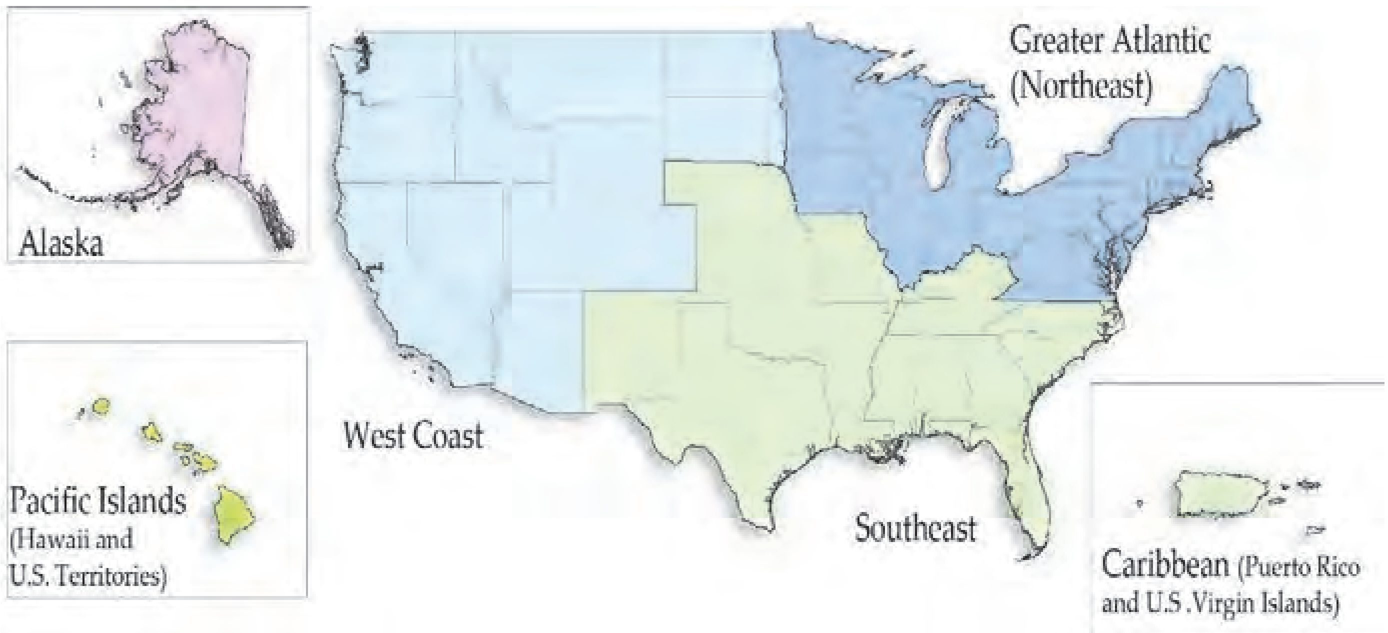
Use the Information for Planning and Consultation tool (IPaC, https://ecos.fws.gov/ipac/) to identify whether ESA-protected species and critical habitat under jurisdiction of USFWS may be present or affected by work in your application area. While IPaC provides valuable information, in many cases it is still beneficial and, for federal agency actions, necessary (unless otherwise specified in your IPaC documents) to contact USFWS offices directly (https://www.fws.gov/offices/index.html). For example, while IPaC may provide project design recommendations and conservation measures that are likely to reduce the potential impacts of proposed activities, USFWS staff can provide additional and more specific recommendations for your particular project.

US Fish and Wildlife Service Regions:

Region 1 – Pacific: http://www.fws.gov/pacific/ecoservices/endangered/
Region 2 – Southwest: http://www.fws.gov/southwest/es/
Region 3 – Midwest: http://www.fws.gov/midwest/endangered/
Region 4 – Southeast: http://www.fws.gov/southeast/endangered-species-act/
Region 5 – Northeast: http://www.fws.gov/northeast/endangered/
Region 6 – Mountain-Prairie: https://www.fws.gov/mountain-prairie/es/endangered.php
Region 7 – Alaska: http://alaska.fws.gov/fisheries/endangered/
Region 8 – Pacific Southwest: http://www.fws.gov/cno/

Are NOAA Fisheries ESA-listed species or designated critical habitat present?

Contact the local regional NOAA Fisheries office to find out whether ESA-protected species or habitat under NOAA fisheries' jurisdiction occurs in your application area. The NOAA Fisheries' species and habitat protected under the ESA occur in marine and fresh waters of coastal states, the Snake River Basin of Idaho, and marine waters of the Pacific and Caribbean islands.



NOAA Fisheries Regions:

Alaska: https://www.fisheries.noaa.gov/alaska/marine-mammal-protection/protecting-marine-life-alaska (marine mammals, sea turtles)
Pacific Islands: https://www.fisheries.noaa.gov/pacific-islands/marine-mammal-protection/protecting-marineanimals-pacific (marine mammals, marine fish, coral, sea turtles)
West Coast: https://www.fisheries.noaa.gov/west-coast/about-us/protecting-marine-life-west-coast (Pacific salmonids and other anadromous species in marine and inland waters, marine mammals, marine fish, abalone, sea turtles)
Greater Atlantic Region: https://www.fisheries.noaa.gov/new-england-mid-atlantic/marine-mammal-protection/protecting-marine-life-new-england-mid-atlantic (Atlantic salmon in marine waters, anadromous sturgeon in marine waters and major rivers, marine mammals, marine fish, sea turtles)
Southeast Region and Caribbean: https://www.fisheries.noaa.gov/southeast/endangered-species-conservation/ esa-section-7-interagency-consultation-southeast-united-states (anadromous sturgeon in marine waters and major rivers, marine mammals, marine fish, coral, sea turtles)

3.4 Cultural and Physical Control of Aquatic Weeds

Marc D. Bellaud: SOLitude Lake Management, Shrewsbury MA; mbellaud@solitudelake.com

Introduction

Methods for cultural and physical control of aquatic weeds are often viewed as strategies that can be readily employed by lake users as well as lake managers. Cultural control typically focuses on education and preventing invasive species introductions from occurring in the first place. Physical control methods are usually non-chemical, non-motorized techniques that are employed to control aquatic weeds and range from hand-pulling to water-level drawdowns, or efforts to alter water or sediment characteristics where weeds are found. As awareness of aquatic nuisance species has grown in recent years, so have efforts to incorporate cultural and physical control methods as important elements of Integrated Weed Management Programs.

Prevention

Many states have prepared official lists of invasive aquatic species and some have even passed legislation to ban their transport or introduction. However, there are often limited resources or mechanisms to enforce rules and prevention efforts are often left to individual lake associations or other volunteer groups. The first step in prevention is regular monitoring to look for new or pioneer infestations. Volunteers can be trained to participate in lake monitoring or "weed-watcher" programs to accurately identify invasive species. In many cases a step-by-step reporting protocol is provided if a new "find" is discovered.

Education is a key component of prevention. Educating lake users and the general public about the threat of invasive species is necessary to prevent new infestations and to sustain effective aquatic plant management programs. Education involves creating public awareness of the problem and familiarizing people with possible solutions. Volunteer labor and public participation are paramount to successful education efforts.

Boat ramp monitoring programs are used to inspect boats and trailers for the presence of invasive species. These are largely volunteer or summer intern positions that try to staff boat ramps during peak use periods. Inspections can either be mandatory or voluntary and usually only take a matter of minutes. Several northeastern states provide annual reports about the number of "saves", which occur when an invasive species is found on a boat or trailer and is removed before the boat is launched. The interaction with boat ramp monitors also provides an opportunity to distribute educational material and conduct surveys about boating habitats and other water bodies that were recently visited.

Boat washing stations are also used at some locations as an aggressive education and prevention measure. Boats and trailers are washed prior to entering and sometimes after leaving a lake. Most aquatic plant fragments capable of surviving out of water are easily seen and can be removed by hand. Washing stations are probably better suited to removing microscopic threats such as zebra mussel veligers, didymo or spiny water flea. Primary considerations for boat washing stations are whether space and utilities for a station are available, the cost of installation, staffing and how wash water is captured and treated. Boaters are sometimes reluctant to utilize volunteer boat washing stations or those where a fee is charged and this is another hurdle that must be overcome for wash stations to be effective.

Assessment and monitoring

The accurate identification of aquatic weed infestations and their associated problems are the first steps toward developing and implementing an aquatic plant management program. Once a program is implemented, monitoring is usually warranted to evaluate the effectiveness of techniques used and to make adjustments in future years. Compliance monitoring and reporting are often a permit requirement and may focus on changes to nontarget species and water quality. The basic protocol that is recommended when initiating an aquatic plant management program is outlined in detail in Section 3.2.

Physical control practices

Aeration or artificial circulation uses electric or solar powered mixers, fountains or compressed air diffuser systems to circulate and add oxygen to the water. The premise is that the addition of oxygen will reduce the amount of available phosphorus and result in less algae growth. The physical circulation or destratification (mixing) of water can also prevent noxious algal blooms from developing (Section 2.18). Benefits of aeration have been clearly documented in all types of water bodies from small, shallow ponds to large, thermally stratified lakes that are using hypolimnetic (deep water) aeration systems. Growth of some aquatic plants appears to be limited by disturbance of the physical water surface and may prevent canopy formation by floating plants such as duckweed or watermeal (Section 2.14). Claims that water circulators control invasive submersed species are unsubstantiated.

Benthic barriers or bottom weed barriers are used for localized control of aquatic plants through compression and by blocking sunlight. Barriers specifically manufactured for aquatic weed control are usually made from materials that are heavier than water such as PVC, fiberglass and nylon. Other fabrics used in landscaping and construction have also been tried. Barriers are usually anchored in place with a variety of fastening pins or anchoring devices. Some of the most common anchors being used are lengths of steel rebar encased in capped PVC pipes, which eliminates any sharp edges that could tear the barriers or be hazardous to swimmers. Sand bags, bricks and steel pins are also commonly used as anchors. Larger panels that are installed in water depths of greater than 4 feet usually require SCUBA divers for proper installation. Several different mechanisms have been devised to unroll the barriers in place during the installation process. Solid fabric barriers often need to be cut or vented to allow gasses to escape and to prevent billowing.

Benthic barriers are usually used to control dense, pioneer infestations of an invasive species or as a maintenance weed control strategy around boat docks and swimming areas. Large installations (greater than one acre) are often impractical due to the high cost associated with purchasing, installing and maintaining the barrier. Benthic barriers should be left in place for a minimum of 1 to 2 months to ensure that target plants are controlled, but barriers must be regularly removed and cleaned of silt; otherwise plants may begin to root on top of or through the barriers. Removal, cleaning and re-deployment is usually required every 1 to 3 years depending on the rate of silt accumulation. Some lakes with volunteer divers have attached barriers to lightweight frames that facilitate rapid deployment and retrieval. Barriers non-selectively control aquatic vegetation and may impact fish and other benthic organisms, which is another reason they are usually used for small localized areas. Many states require permits for the use of benthic barriers.

Drawdown or the lowering of the water level can be used to effectively control a number of invasive submersed species. This technique is used mostly in the northern US to expose targeted plants to freezing and drying conditions. Water is either gravity drained using a low-level gate valve or through a removable flashboard system on a dam. Siphoning or pumping can also be performed in lakes with insufficient outlet structures. A principal attraction of drawdown is that it is typically an inexpensive weed control strategy for lakes with a suitable outlet structure. Annual drawdown programs can result in sediment compaction and changes in substrate composition. Drawdowns are also utilized to provide protection from ice damage to docks and other shoreline structures and to allow for shoreline clean-up and repairs by lake residents.

Plants that are usually controlled by drawdowns include many submersed species that reproduce primarily through vegetative means such as root structures and vegetative fragmentation. Some invasive submersed species most commonly targeted by drawdown include Eurasian watermilfoil (Section 2.3), variable watermilfoil (*Myriophyllum heterophyllum*), fanwort (Section 2.6), egeria or Brazilian elodea (Section 2.5) and native coontail (*Ceratophyllum demersum*).

Waterlilies (*Nymphaea* sp.) can also be effectively controlled, provided sediments can be sufficiently dewatered to allow for the freezing and drying conditions required to control this species. Seeds and other non-vegetative propagules such as turions or winter buds are not controlled by drawdown; in fact, species that reproduce by these means may actually increase following drawdown programs. Many species of pondweed (*Potamogeton* spp.) have increased following drawdown programs and highly opportunistic species like hydrilla (Section 2.2) may expand rapidly following drawdown.

A general rule of thumb is to maintain drawdown conditions for 6 to 8 weeks to ensure sufficient exposure to freezing and drying conditions. Excessive snow cover or precipitation can limit the effectiveness of this technique. Drawdowns are usually timed to begin during the fall months to avoid stranding amphibians, molluscs and other benthic organisms with limited mobility. Care must also be taken to leave enough water to support fish populations and avoid impacts during key spawning periods. Drawdowns can have negative impacts on desirable aquatic plant species. Drawdowns can also impact adjacent wells and wetlands, so it is important to know the downstream channel configuration, capacity and flow requirements. When properly utilized, drawdowns can be a low-cost or no-cost strategy to incorporate into an integrated management program. Many states require permits for drawdown programs.

Hand pulling or hand harvesting is one of the simplest and most widely used methods to control aquatic weed growth and can be performed by wading or from a small boat in shallow water. Snorkeling equipment, surface supplied air systems or SCUBA divers are often used in water greater than 4 to 5 feet deep and for more intensive hand pulling programs. Boat or barge mounted suction pumps fitted with straining systems are often used to increase removal efficiency and are commonly referred to as Diver Assisted Suction Harvesting or DASH. Hand pulling can be a highly selective technique, provided the target species can be easily identified, and is usually used as a component of invasive species management programs to target new infestations with low plant density (generally less than 500 stems per acre). Hand pulling can be used to remove more dense plant growth over small areas, but benthic barriers or suction harvesting may be more effective approaches in these situations. It is often an important follow-up strategy to a herbicide treatment program to extend the duration of plant control.

When hand pulling a plant like Eurasian watermilfoil, the roots should be carefully dislodged from the bottom substrate so that the entire plant can be collected and removed to prevent vegetative regrowth. Once the bottom substrate is disturbed, suspended sediment often greatly reduces visibility, which results in the need to make multiple passes over the same area. In larger hand pulling programs that use multiple divers or DASH equipment, it is often advantageous to have people in boats that can collect dive bags full of weeds and can try to capture escaping plant fragments using pool skimmers. Waterchestnut (Section 2.10) is a noxious invasive species that has been effectively managed in several locations by hand pulling programs. This floating-leaved plant is easily identified and is a true annual plant that usually drops its seeds in late summer. Hand pulling efforts are usually performed for several weeks during the summer months before seed drop occurs. Several successful volunteer waterchestnut hand pulling programs have been organized and implemented in the Northeast.

Hand rakes of varying sizes and configurations are manufactured and sold for aquatic weed control. Many of these hand rakes are lightweight aluminum, with rope tethers that are designed to be thrown out into a swim area and dragged back onto shore. Some are designed to cut the weeds instead of raking them back to shore. While these may be cost-effective strategies to manage individual swim areas, there is a risk that these rakes will make the problem worse by creating weed fragments that can escape and infest other portions of the lake.

Nutrient inactivation involves the application of aluminum (as aluminum sulfate), iron salts, calcium compounds (lime) or rare-earth metals like lanthanum to remove phosphorus from the water column and to inactivate phosphorus in the sediment. Removing and inactivating phosphorus can effectively discourage algal blooms from developing, but the growth of most rooted vascular plants is usually limited by nitrogen and there are no compounds readily available that bind with nitrogen in the sediment. Injecting sediments with alum and lime has been attempted, but suppression of vascular plant growth was not significant. Nutrient inactivation remains best suited for water quality improvement and algal control. In fact, reducing water column nutrients and algae may encourage even more dense infestations of nuisance rooted plants due to improved water clarity and light penetration, which may allow weeds to grow in deeper areas.

Shading through the use of EPA-registered dyes or surface covers attempts to limit light penetration and restrict the depth at which rooted plants can grow. Dyes are usually considered non-toxic solutions that give the water a blue or black color. The use of dyes is often limited to smaller golf courses or ornamental ponds because they make the water appear artificial. Dyes have little use in larger water bodies; in addition, if the pond or lake has a flowing outlet, multiple treatments will likely be required and state or local permitting may be required. Surface covers made from various fabrics or plastic materials can be used to prevent light penetration and control rooted plant growth. This approach is generally not used in recreational ponds and lakes since they would impair access to or use of the lake. Recent studies have shown that this can be an effective means of controlling plants that do not produce seeds or other vegetative reproductive propagules, but its application is usually limited to small, highly controlled areas.

Weed rollers consist of a roller on the lake bottom that is powered by an electric motor and travels forward and reverse in up to a 270-degree arc around a pivot point. Rollers can be up to 30 feet long and are typically installed at the end of a dock. Plants initially become wrapped around the roller and are dislodged from the sediment; the constant motion of the rollers then disrupts and compresses the bottom sediments, which prevents plants from becoming reestablished. Because the rollers travel along a pivot point, they reportedly can be used in several different substrate types. Weed rollers are only practical for managing small areas. They may disrupt fish spawning or other benthic organisms, but these impacts would likely be minimal or highly localized. Many states require permits for the use of weed rollers.

Summary

There are a number of cultural or physical methods that can be employed by lake associations or individual lakefront owners to control aquatic weeds, but the most important function of stakeholders is to develop a prevention plan. The vast majority of new weed infestations are found near boat ramps, so these areas should be surveyed on a regular basis. Residents that regularly spend time on the lake should obtain plant identification materials from state agencies or other information sources so that exotic plants can be accurately identified and targeted for treatment. Management plans should be developed for rapid response; in other words, plans should be developed proactively and stakeholders shouldn’t wait for an invasive species to appear before creating a plan. Prevention and rapid response should be top priorities among lake associations because these are the most cost-effective and ecologically sound means of protecting aquatic resources from invasive species.

Photo and illustration credits:

Page 129: Boat wash station; William Haller, University of Florida
Page 130: Benthic barrier; William Haller, University of Florida
Page 131: Drawdown; University of Florida Center for Aquatic and Invasive Plants (photographer unknown)

3.5 Mechanical Control of Aquatic Weeds

William T. Haller: University of Florida, Gainesville FL (emeritus); whaller@ufl.edu

Introduction

The term "mechanical control" as used in this chapter refers to control methods that utilize large power-driven equipment. The simplest method of mechanical control might be the dragging of an old bedspring or other heavy object behind a boat to rip up and remove submersed weeds from a beach used for swimming. Mechanical control has been practiced in the US for over a century and almost every engineer has a conceptual idea of how to build the "perfect aquatic weed harvester." One major obstacle to designing a universal mechanical harvester is the diversity of plants and environments where the equipment will be employed. This has led to the development – and ultimate abandonment – of a plethora of various types of equipment throughout the years. Primary factors to be considered when selecting a mechanical control method are the types of weeds to be controlled and the habitats they occupy.

Wetland or emergent weeds

Wetland habitats are typical marsh ecosystems with periodically inundated soils, a high water table and/or water depths of up to two feet. Emergent plants such as phragmites (Section 2.15), purple loosestrife (Section 2.16), native cattails (*Typha* spp.) and other wetland plants are common in these areas. Mechanical control is employed on a very limited basis in these "protected" habitats because access is often difficult and the destruction and alteration of protected wetlands in the US is highly regulated.

While there is very limited mechanical weed control conducted in wetlands, the mechanical control method most commonly employed by land managers is mowing. For example, dense stands of phragmites may be mowed during dry seasons or under drought conditions to provide temporary control. Also, chain saws and hand-pulling have been used in wetlands of southern Florida for control of melaleuca (*Melaleuca quinquenervia*) trees and seedlings, respectively. Ducks Unlimited and other resource agencies have used dredges and choppers of various types to reclaim or restore wetlands, but the primary purpose of these activities is not solely weed control. Overall, mechanical weed control is rarely used for invasive species management in wetlands and shallow-water areas due to the likelihood of creating significant environmental damage.

Floating weeds

Most mechanical weed control occurs in water greater than 2 feet deep and the type of plant to be controlled (floating or submersed) must be taken into consideration when selecting a mechanical control method. Floating plants should be evaluated separately from submersed plants because floating plants produce 10 to 20 times more biomass than submersed plants – biomass that has to be chopped, picked up or otherwise moved away from the harvesting site. For example, the standing crop or biomass of an acre of undisturbed waterhyacinth (Section 2.11) can weigh 200 to 300 tons per acre, whereas an acre of hydrilla (Section 2.2) or Eurasian watermilfoil (Section 2.3) may only weigh 10 tons or less per acre. Most mechanical harvesters are able to pick up and transport less than 5 tons of biomass per load, so there is a huge difference in the time, effort and expense required to mechanically harvest floating plants compared to submersed aquatic weeds.

Two additional problems associated with floating plants are their ability to move by wind or water currents and their location in lakes and rivers. For example, there may be only one access point where plants can be loaded onto trucks for disposal. Plants may initially be located close to the work site, but on another day – after a change in wind direction – plants may be on the other side of the lake and will need to be transported a long distance before they can be off-loaded. Also, floating plants are often blown into shallow waters along shorelines, which may be lined with cypress or willow trees. Most harvesters cannot work in water less than 2 feet deep and cannot navigate in and among trees, rocks or stump-fields in flooded reservoirs.

Submersed weeds

Mechanical harvesting of submersed weeds, primarily curlyleaf pondweed (Section 2.4) and Eurasian watermilfoil, has been widely used in the Northeast and Midwest. The shallow shores of even very deep lakes in these regions often support the growth of these submersed weeds and multiple harvests provide control during the recreational season. Governmental entities (including state, county and local governments) have subsidized weed removal from public lakes in some locations to maintain high use areas and to promote tourism and general utilization of the water resource. In other areas, lake associations and groups of homeowners often hire aquatic management companies for weed removal services. Although mechanical harvesting is often used in northern lakes to control submersed weeds, this method has less utility in southern states due to longer growing seasons and much larger-scale coverage of weeds in the shallow lakes and reservoirs more commonly encountered in the Southeast.

Examples of mechanical equipment

Cutter boats have been used in the US in one form or another for decades. For example, a small barge with a steam engine powered an underwater sickle bar mower in the Upper Chesapeake Bay/Potomac River area at the turn of the century. Submersed plants cut by the barge floated from the harvested area via river and tidal currents. Also, the US Army Corps of Engineers built sawboats in the early 1900s for use in navigable waters of Louisiana and Florida. These boats had gangs of circular saws mounted about an inch apart on a spinning shaft that was mounted at the bow of the boat and only penetrated the top inch or two of the water. These sawboats chopped up waterhyacinth, alligatorweed (*Alternanthera philoxeroides*) and grasses which formed intertwined mats of floating vegetation. The chopped vegetation was allowed to flow downstream or to salt water. Cutter boats have been used more recently to clear navigation channels, but this equipment is not usually used in lakes and non-flowing systems because most cut weeds float and survive for long periods of time. Fragments such as these can establish in other parts of the water body or wash up on swimming beaches. Cutter boats create large amounts of fragments and vegetative cuttings, so the ability of the target weed to spread and grow from fragments should be evaluated before cutter boats are employed as a primary mechanical control method.

Shredding boats are used to control emergent and floating plants. The most common type of shredder is the "cookie cutter," which consists of two spinning blades (3 to 4 feet wide) that are mounted behind a steel hood on the front of a small but powerful barge. The boat is propelled by hydraulically raising and lowering the blades and changing the direction of the blades (see https://texasaquaticharvesting.com/). Recently, bow mounted high-speed flail mower blades have been tested for chopping and shredding floating and emergent plants. Similar to other mechanical control equipment, shredder boats are very specialized pieces of equipment, are non-selective and create many plant fragments. However, they work well when used in the areas for which they are designed and are frequently used in wetland restoration projects, where removal of cut vegetation is too expensive or not feasible.

Rotovators are highly specialized large aquatic rototillers. The rotovator head is lowered onto the lake or river bottom and "tills" the sediments, which chops up and cuts loose submersed plants. A floating boom is usually placed around the work area while the rotovator spins on the lake bottom; uprooted plants float to the surface and are removed from along the barrier by hand or mechanical means. Rotovators have been used mostly in the Pacific Northwest, where the submersed weed Eurasian watermilfoil grows in rocky bottom areas and roots in the shallow soil between and among small rocks. The rotovator head moves the rocks around and uproots the weeds from the shallow soils and rock crevasses.

Dredges are not usually used for aquatic weed control due to high costs associated with their operation, but weed control can be a benefit of dredging that is done for other reasons. Shallow ponds and lakes that have filled with silt and organic matter over time may only be 3 to 4 feet deep and provide an ideal environment for excessive growth of submersed weeds and native plants such as cattail and waterlily. If the water depth of the pond is increased to 6 to 10 feet by dredging, it is unlikely that emergent plants such as cattail will continue to grow. However, submersed weeds will almost certainly still infest the pond if water depth and clarity requirements for growth of the weeds are met.

Cut and removal harvesters are the most widely used types of equipment employed for mechanical control in the US. The first machines were developed in the 1950s by a Wisconsin company to harvest Eurasian watermilfoil and curlyleaf pondweed from the edges of the hundreds of lakes in the Upper Midwest. These lakes are generally deep in the middle and aquatic weeds naturally grow in the shallow littoral areas, which receive intensive use for swimming and docking over the 3 to 4 month summer season. These harvesters are powered by side-mounted paddle wheels which operate independently in forward or reverse. As a result, they are highly maneuverable around docks and boat houses. Also, the machines can operate in as little as 12 to 18 inches of water. These harvesters typically cut plants off at depths to 5 feet deep and in swaths 8 feet wide with a hydraulically operated cutter head and convey the cut plants into a storage bay on the harvester. When the harvester is full, it offloads harvested plants onto a transport barge by conveyer belts and the transporter takes the vegetation to shore, where it is dropped onto a conveyor to elevate the load to a truck for disposal. If you have read this carefully, you have counted four pieces of equipment: a harvester, a transporter, a shore conveyer and a truck. All this equipment may not be necessary, as mechanical harvesting is obviously tailored to a particular situation and is very site-specific. Also, some harvester trailers have been modified to allow them to transport cut weeds to the disposal site. This system or a setup with similar equipment has been used for 50 years in lakes from New England to California, but is mostly employed in northern lakes where one or two harvests during spring and summer can provide nearly weed-free conditions for the seasonal summer use of these lakes.

Advantages and disadvantages

There are many advantages to mechanical harvesting. These include:

- Water can be used immediately following treatment. Some aquatic herbicides have restrictions on use of treated water for drinking and irrigation. Also, plants are removed during mechanical harvesting and do not decompose slowly in the water column as they do after herbicide application. In addition, oxygen content of the water is generally not affected by mechanical harvesting, although turbidity and water quality may be affected in the short term.
- Nutrient removal is usually insignificant because only small areas of lakes (1 to 2%) are typically harvested; however, some nutrients are removed with the harvested vegetation. It has been estimated that aquatic plants contain less than 30% of the annual nutrient loading that occurs in lakes, but this value varies widely between lakes and their water catchment areas.

- The habitat remains intact because most harvesters do not remove submersed plants all the way to the lake bottom. Like mowing a lawn, clipped plants remain rooted in the sediment and regrowth begins soon after the harvesting operation.

- Mechanical harvesting is site-specific because plants are removed only where the harvester operates. If a neighbor wants vegetation to remain along their lakefront, there is no movement of herbicides out of the intended treatment area to impact the neighbor's site.

- Herbicide concerns remain widespread despite extensive research and much-improved application, use and registration requirements that are enforced by regulatory agencies (Section 3.7). Mechanical harvesting, despite some environmental concerns (as outlined below), is perceived to be environmentally neutral by the public.

- Utilization of harvested biomass is thought by many to be a means of offsetting the relatively high costs and energy requirements associated with mechanical harvesting. Unfortunately, no cost-effective uses of harvested vegetation have been developed, despite much research examining the utility of harvested plant material as a biofuel, cattle feed, soil amendment, mulch or even as a papermaking substrate. As much as 95% of the biomass of aquatic plants is water, so 5 tons of Eurasian watermilfoil yields only 500 pounds of dry matter. In addition, cut plants in northern lakes are only available for 3 to 4 months of the year.

The easiest way to highlight the disadvantages of mechanical harvesting is to point out that major producers of farm equipment (for example, John Deere or New Holland) do not mass-produce equipment designed for the mechanical harvesting of aquatic weeds. Farmers are famous for efficiently cutting, harvesting and moving hay, corn and grain crops; they constitute a large market and specialized equipment is available to them. On the other hand, the demand for aquatic weed harvesters is very small, so the equipment associated with these operations is often custom-made and expensive. Other disadvantages include:

• The area that can be harvested in a day depends on the size of the harvester, transport time, distance to the disposal site and density of the weeds being harvested. These factors can result in a wide range of costs. The cost of harvesting is site-specific, but mechanical harvesting is generally more expensive than other weed control methods due to the variables noted above and the generally high capital outlay required to purchase equipment that may only be used for 3 or 4 months per year.

• Mechanical harvesters are not selective and remove native vegetation along with target weeds. However, this is probably not a significant disadvantage since native plants and weeds will likely return by the next growing season, if not sooner.

• By-catch, or the harvesting of nontarget organisms such as fish, crayfish, snails and frogs along with weeds, may be more of a concern, but the degree or extent of harvesting should be considered. Research on fish catch during mechanical harvesting of submersed vegetation has shown that 15 to 30% of some species of fish can be removed with cut vegetation during a single harvest. If the total area of a lake that is harvested is 1, 5 or 10% of the lake's area, this will likely be of little consequence. However, if the management plan for a 10-acre pond calls for complete harvests 3 times per year, then the issue of by-catch of fish deserves more consideration.

• Regrowth of cut vegetation can occur quickly. For example, if hydrilla can grow 1 inch per day as reported, a harvest that cuts 5 feet deep could result in plants reaching the water surface again only two months after harvesting. Speed of regrowth depends on the target weed, time of year harvested, water clarity, water temperature and other factors.

• Floating plant fragments produced during mechanical harvesting can be a concern because most aquatic weeds can regrow vegetatively from even small pieces of vegetation. If an initial infestation of aquatic weeds is located at a boat ramp, care should be taken to minimize the spread of fragments to uninfested areas of the lake by maintaining a containment barrier around the area where mechanical harvesting will take place. On the other hand, if a lake is already heavily infested with a weed, it is unlikely that additional fragments will spread the weeds further. However, homeowners downwind of the harvesting site may not appreciate having to regularly rake weeds and floating fragments off their beaches.

• Disposal of harvested vegetation can be an expensive and difficult problem after mechanical harvesting. Research during a project in the 1970s on Orange Lake in Florida compared the costs of in-lake disposal to the transport, off-loading and disposal of cut material at an upland site. As water levels on Orange Lake decreased during a drought period, the mechanical harvester was allowed to off-load cut vegetation along the shoreline among emergent vegetation instead of transporting harvested plants to the shore for disposal. The cost of in-lake disposal reduced the per-acre cost by about half when compared to transporting the vegetation to shore, loading it into a truck and disposing of the plant material in an old farm field.

• Some lakes or rivers may not be suitable for mechanical harvesting. If there is only one public boat ramp on a lake and it is not close to the area to be harvested, the costs of moving the cut vegetation from the harvester to shore will add significantly to the cost of the operation. Harvesters are not high-speed machines and move at 3 to 4 mph, so if a river flows at 2 mph and the harvester has to travel upstream to the off-loading site, well, do the math! Off-loading sites usually must have paved or concrete surfaces because the weeds are wet and an unpaved off-loading site can quickly become a quagmire.

Recent advances in deep-water harvesting

The rising cost of herbicides for hydrilla control and the development of fluridone-resistant populations of hydrilla in Florida lakes served as the stimulus for a multi-agency evaluation of improved mechanical control techniques for hydrilla control during the early 2010s (see http://www.fapms.org/aquatics/issues/2012fall.pdf). Improved efficiency and reduced costs associated with mechanical harvesting would likely result if: 1) harvesters were larger, since this would reduce the number of trips and transport time needed to offload harvested material; and 2) deeper harvesting was possible, since this would reduce weed biomass in deeper waters, resulting in longer intervals between harvesting events because weeds would require more time to regrow to problematic levels.

To test this theory, a 70-foot harvester with a cutter head capable of harvesting weeds in water as deep as 10 feet was developed by a commercial firm. The harvester was also equipped with a GPS tracking unit to allow the operator to harvest plants that are difficult or impossible to see from the operator’s position on the harvester. This equipment was used in an early-season (March) operation to remove hydrilla that was 3 to 6 feet tall and growing in 8 to 9 feet of water and allowed harvesting of 2 to 4 acres per hour. Topped-out or surface-matted hydrilla can weigh as much as 14 tons (fresh weight) per acre, but this low-growing hydrilla, which was harvested from the soil line up through the water column, averaged only 1,000 pounds per acre. Fish by-catch was greatly reduced during this operation compared to previous operations in surface-matted hydrilla because: 1) oxygen levels were consistent throughout the water column due to less dense weed populations than those encountered later in the season, so fish did not preferentially inhabit weed beds; and 2) the relatively low weed density entangled fewer fish, so fish could escape the harvester. This early-season deep-water harvesting operation had a negligible impact on water quality because the harvester’s propulsion unit was farther away from the lake bottom and sediments were not disturbed as they would be in shallower water. In addition, control was achieved for 5 to 7 months; this is a decided improvement over the 2 to 3 months of control provided by shallow-water harvesting and could allow longer intervals between harvesting, which would significantly reduce the costs associated with mechanical harvesting. Additional trials are planned, but it appears that the strategy of using a GPS-assisted larger harvester to manage less dense weed infestations in deeper water may significantly increase the efficiency of mechanical harvesting of submersed weeds.

Summary

This discussion is not intended to include all the machines that are available for mechanical control of aquatic weeds and it is likely that new ideas and equipment will be developed as time passes. It is important to remember that each site and each weed has characteristics that may require a particular type of mechanical harvester and may preclude the use of other mechanical methods of control. There is a vast repository of information available on the internet and the best source of information is the conservation or regulatory agency in your state. In fact, most states require that permits for mechanical harvesting be obtained before work can begin.

Photo and illustration credits:

Page 133: Mower; William Haller, University of Florida
Page 135: Cutter boat; William Haller, University of Florida
Page 136: Harvester; William Haller, University of Florida
Page 137: Conveyor; Jeff Schardt, Florida Fish and Wildlife Conservation Commission

3.6 Introduction to Biological Control of Aquatic Weeds

James P. Cuda: University of Florida, Gainesville FL; jcuda@ufl.edu

Introduction

There are many herbivores or plant-eating animals in the aquatic environment, including manatees, moose, muskrat, turtles, fish, crayfish, snails and waterfowl. These animals are general herbivores and may prefer to eat certain types of plants, but do not rely on a single plant species as a primary food source. Although these animals do consume aquatic plants and therefore reduce the growth of some species, they generally do not have a significant impact on overall plant growth because they feed on many different plants and are not considered biological control agents. Biological control (also called biocontrol) is broadly defined as the planned use of one organism (for example, an insect) to control or suppress the growth of another organism such as a weedy plant species. Biocontrol of weeds is primarily the search for, and introduction of, species-specific organisms that selectively attack a single target species such as an exotic weed. These organisms may be insects, animals or pathogens that cause plant diseases, but most biocontrol agents are insects. Biocontrol has been studied and used for more than a century and has developed into a complicated and technical science based on a number of principles that will be discussed in this chapter. Two different approaches are currently used in the biocontrol of aquatic weeds: classical (importation) and non-classical (augmentation, conservation).

Classical biocontrol is by far the most common biological control method and typically involves the introduction of natural enemies from their native range to control a nonnative invasive plant. The excessive growth of a weed in its new habitat is due in part to the absence of natural enemies that normally limit or slow the growth, reproduction and spread of the weed in its native range. Successful classical biocontrol seeks to reunite an invasive plant with one or more of its coevolved natural enemies to provide selective control of the weed. Thus, classical biocontrol can be defined as the planned introduction and release of nonnative target-specific organisms (usually arthropods, nematodes or plant pathogens) from the weed's native range to reduce the vigor, reproductive capacity or density of the target weed in its adventive (new or introduced) range.

Classical biocontrol offers several advantages over other weed control methods. It is relatively inexpensive to develop and can be incorporated with other methods of weed control. Classical biocontrol provides selective, long-term control of the target weed and because biocontrol agents reproduce, they will usually spread on their own throughout the infested area. Some of the strengths of classical biocontrol also contribute to its shortcomings. For example, it may not be possible to find a biocontrol agent that effectively controls a single weed and selectively attacks only that particular weed. When potential biocontrol agents are identified, their establishment and suppression of the target weed in the introduced area are not guaranteed. Even if biocontrol agents do successfully establish in their introduced areas, control is not immediate and agents may require many years to have a major impact on target weeds. Finally, once a biocontrol agent is established, it cannot be recalled if desirable nontarget species are affected by the agent.

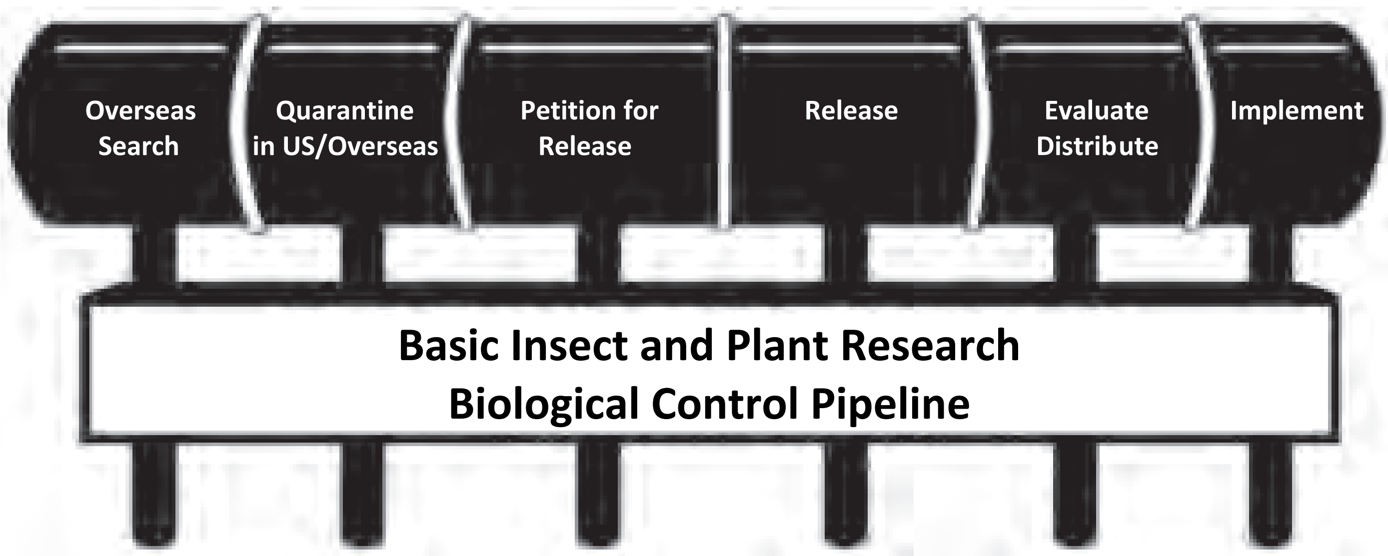
Non-classical biocontrol involves the mass rearing and periodic release of resident or naturalized nonnative aquatic weed biocontrol agents to increase their effectiveness. Savvy home gardeners employ this approach when they purchase ladybird beetles to control aphids (insects that are serious pests of fruits, vegetables and ornamentals) in their home gardens. Augmentative or repeated releases of native or naturalized insects have occasionally been used for suppression of alligatorweed (*Alternanthera philoxeroides*), waterhyacinth (Section 2.11), hydrilla (Section 2.2) and Eurasian watermilfoil (Section 2.3).

The "new association" approach is a variation of classical biocontrol. **New association biocontrol** differs from classical biocontrol in that the natural enemies or biocontrol agents have not played a major role in the evolutionary history of the host plant and are therefore considered new associates. Because organisms used in the new association approach are not entirely host-specific, this approach is appropriate only in cases where the target weed has few or no closely related native relatives in the area of introduction.

A good example of the new association approach is the milfoil weevil (*Eurychiopsis lecontei*), which is native to North America and attacks native species of milfoil (*Myriophyllum* spp.) in the US and Canada. Recent studies have shown that milfoil weevils reared on the introduced weed Eurasian watermilfoil (*Myriophyllum spicatum*) not only develop faster and survive better on the exotic invasive milfoil, but also preferentially attack the nonnative weed species over the native northern watermilfoil (*M. sibiricum*), its natural host plant. This phenomenon was unexpected, unplanned and unusual. Many aquatic resource managers are currently evaluating this natural occurrence to determine how best to include this weevil in weed control programs.

Procedures in a classical weed biocontrol project

Weed biocontrol scientists (most of whom are entomologists or plant pathologists) develop and refine procedures for locating, screening, releasing and evaluating biocontrol agents. All countries currently conducting weed biocontrol projects follow this protocol in one form or another to ensure that candidate organisms are safe to introduce. The normal process in a classical biocontrol program is often referred to as the "pipeline." The pipeline consists of the following series of well-defined steps:



• Step 1: Target selection. Ideal targets for biocontrol are invasive nonnative plants with no closely related native plants in their introduced ranges. Scientists read the literature associated with the target weed to learn where the weed came from (geographic origin), what desirable plants are closely related to the weed and to identify potential natural enemies. Some pathogens and insect herbivores often exhibit what Harley and Forno (1992) refer to as "fine tuned" adaptation to specific populations and genotypes of their host plants. Molecular tools such genotyping by sequencing (GBS) and comparative genomics can be used to match the target weed precisely with its co-evolved natural enemies.

• Step 2: Overseas and domestic surveys. Scientists visit the native range of the target weed to search for natural enemies that may affect and slow the growth of the weed. They evaluate how the target weed is damaged by organisms in its native range to determine if these organisms may be useful as biocontrol agents for the target weed in its introduced range. Another predictor of success is past performance; if a biocontrol agent has been successful in controlling a weed in some countries, there is a high probability that it will be successful in other countries as well. Scientists also conduct surveys in the weed's introduced range (domestic surveys) to avoid introducing biocontrol agents that are already established but ineffective.

• Step 3: Importation and quarantine studies. If an organism attacks only the exotic weed, and not desirable species, scientists request permission from the US Department of Agriculture to import the organism to the US for host range testing. Once permission for importation is granted, the potential biocontrol agent is brought to the US and placed in an approved quarantine laboratory where it cannot escape and is carefully studied to ensure it will not harm desirable species such as crops and native plants.

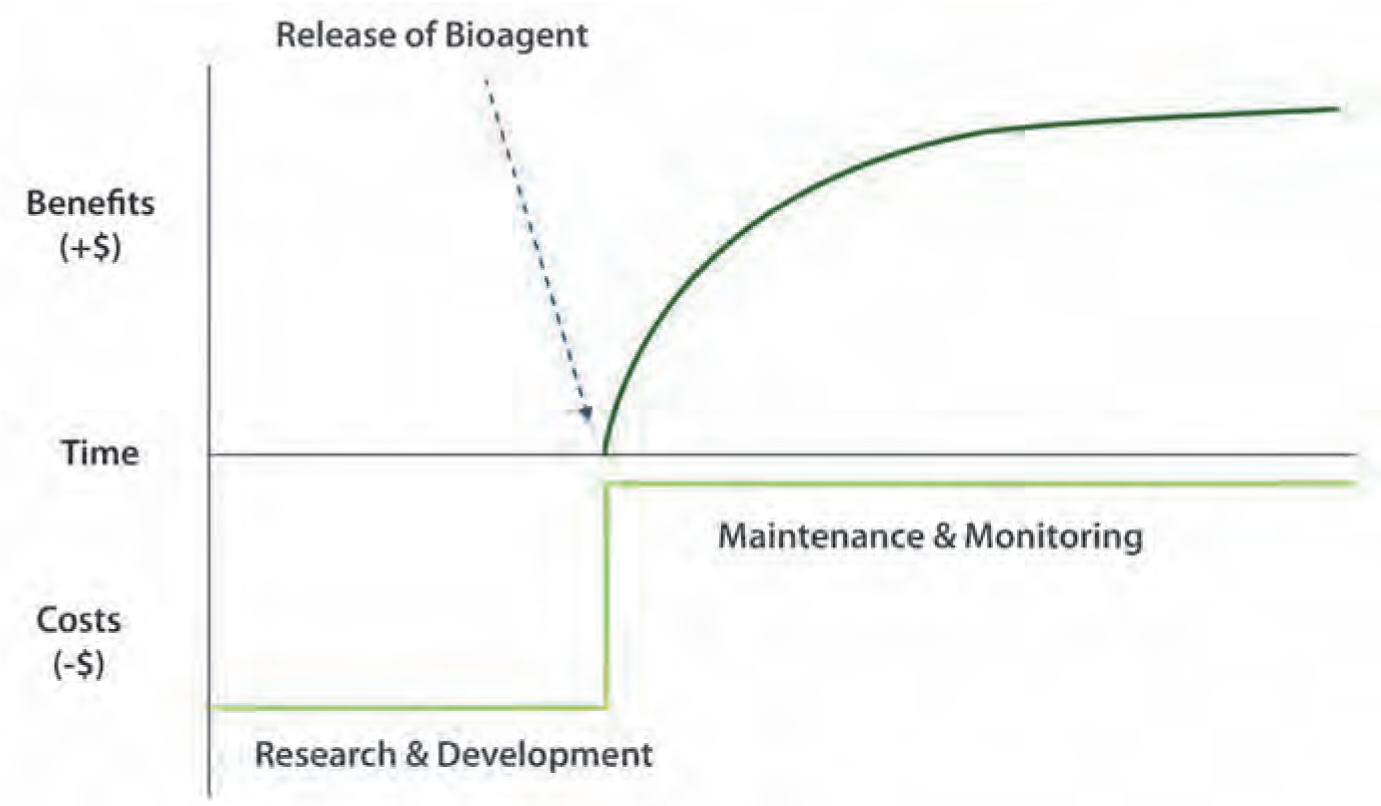
• Step 4: Approval for release. The results of quarantine studies are forwarded to the appropriate federal and state agencies, who determine whether the organism is safe to release. These independent agencies may request that additional testing be done to evaluate the effect of the organism on additional native plants, especially threatened or endangered species, as well as related plants not included in the original quarantine studies.

• Step 5: Release and establishment. Once the biocontrol agent is shown to pose minimal risks to desirable native, ornamental and crop plants, permits are issued and large numbers of the biocontrol agent are reared. This ensures that population densities will be high enough to allow breeding colonies of the agent to establish in the field. Scientists then release the biocontrol agent in multiple locations to increase the likelihood of successful establishment.

• Step 6: Evaluation. Scientists monitor all introduced biocontrol agents after field release to confirm establishment and dispersal of the agent. Multiple releases of the organism may be necessary initially to maintain populations that are adequate for control of the weed species. Additional studies are conducted to determine the effect of the biocontrol agent on the target weed as well as on additional nontarget plants.

• Step 7: Technology transfer. Resource managers are trained in the identification and use of the biocontrol agent. Scientists also collaborate with those using the biocontrol agent to determine the best methods to integrate biocontrol with other weed control methods.

Successful biocontrol programs are expensive at the beginning and can take a long time to develop, but biocontrol can reduce the need for other weed control methods such as herbicides and mechanical harvesting. Because classical biocontrol can provide selective, long-term control of a target weed and biocontrol agents naturally spread by reproducing, the use of biocontrol results in the reduction or elimination of costs for other aquatic weed control methods.



Safety – what has to be done to introduce a biocontrol agent?

Host specificity is fundamental to biological weed control because it ensures that an introduced agent will not damage desirable plants. Host-specific, coevolved natural enemies are considered good candidates for use as biocontrol agents because they are unable to reproduce on plants other than their weedy hosts. In addition, these types of organisms have proven to be the safest to introduce because they are least likely to damage nontarget species. Because host-specific natural enemies reproduce only when they have access to their host plants, their populations are limited by availability and abundance of the target weed.

Potential biocontrol agents are first tested for effectiveness and host specificity in their native range, then promising candidates are brought to quarantine laboratories in the US for final host range testing to determine whether the organism can live and reproduce on native plants. Before scientists can release an agent into the US for classical biocontrol of an invasive aquatic plant, the potential agent must undergo rigorous testing in quarantine to ensure it will only survive on the weed species and will not harm nontarget species. The potential biocontrol agent is offered a series of carefully chosen plants in two different types of tests to determine if the agent is safe to release. In no-choice tests, the agent is given access only to a nontarget plant to determine if it will attack the nontarget plant if the agent's host plant (the target weed) is unavailable. In multiple-choice tests, the agent is offered the target weed and at least one nontarget plant to determine whether the agent damages only the target weed. **Nonnative biocontrol agents can only be released if these tests show that the agent requires the host plant to survive and reproduce and that it will not attack desirable nontarget plants**.

Selecting organisms as candidates for classical biocontrol is a complicated and lengthy process because scientists must identify natural enemies that have developed a high degree of specificity with their weedy host plants. According to established guidelines, no potential biocontrol agent can be introduced into a new environment before its host range is determined. Multiple screening tests are usually required to identify the host range of the agent and scientists must conduct a number of host range tests in the field and laboratory (egg laying, larval development and feeding by adults) to determine whether a biocontrol agent requires the presence of the weedy host plant to survive. Candidate organisms that are able to live and reproduce without access to their weedy host fail the host specificity requirement; they are then dropped from further consideration and quarantined populations are destroyed.

The review process – why does it take so long to release a biological control insect?

The US Department of Agriculture's Animal and Plant Health Inspection Service, Plant Protection Quarantine permitting unit (hereafter referred to as APHIS) is responsible for approving the release of any biocontrol agent in the US. The Plant Protection Act of 2000 gives APHIS the authority to regulate "any enemy, antagonist or competitor used to control a plant pest or noxious weed." Scientists must apply for a permit from APHIS before they can import a potential biocontrol agent into the US for host specificity testing and approved biocontrol agents must be sent directly to a high-security quarantine facility upon entry into the US. There are a number of secure quarantine facilities located throughout the US that are specifically designed and constructed for biocontrol research on aquatic and terrestrial weeds.

After host specificity testing is completed, a permit must be obtained from APHIS before the biocontrol agent is released in the field. A multi-agency Technical Advisory Group for Biological Control Agents of Weeds (TAG) reviews information submitted by the requesting scientist to APHIS. TAG members review test plant lists for weed biocontrol projects, advise weed biocontrol scientists, review petitions for field release of weed biocontrol agents and provide APHIS with recommendations on the proposed release.

In addition to submitting a release petition to TAG and APHIS, scientists contact the Department of the Interior to ensure that threatened and endangered species are included in their test plant list. Release of nonnative weed biocontrol agents also requires compliance with the Endangered Species Act (ESA) (Section 3.3) and the National Environmental Policy Act (NEPA). Scientists must complete an Environmental Assessment (EA) document that outlines the potential impact of the biocontrol agent on the environment in order to comply with the NEPA. The EA provides the public with possible positive and negative environmental impacts that might occur if the new biocontrol agent is released in the US. Scientists must also submit to the US Fish and Wildlife Service a Biological Assessment (BA) document in order to comply with the ESA. The review process is designed in this manner to ensure that there is little chance the introduced biocontrol agents will become pests themselves. Once a weed biocontrol agent is released, several years may be required for the organism to establish and impact the target weed. Scientists continually monitor dispersal of the agent, collect data on its effectiveness to the target weed and also monitor the agent's effect (if any) on nontarget plants during this time.

What is considered a success?

Successful biocontrol of an aquatic weed is a function of the biocontrol agent's capacity to reproduce on individual plants and to build populations large enough to damage the weed's population. However, high population densities of a biocontrol agent do not necessarily guarantee success and effective biocontrol may only occur when the weed is stressed concurrently by local climatic conditions, competing plants or other natural enemies.

In general, insect biocontrol of aquatic weeds in the US has been successful since it was first used to control alligatorweed in 1964. Insects have provided varying levels of control (from complete control to suppression of growth) of the aquatic form of alligatorweed and of waterhyacinth in most areas where insect biocontrol has been attempted. The high success rate achieved by these projects may be correlated with the growth form of the weeds, their susceptibility to disease-causing pathogens, the fluid nature of the aquatic environment, the organisms used as biocontrol agents, or a combination of these factors. For instance, waterhyacinth and the aquatic form of alligatorweed produce floating mats, a growth habit that makes them susceptible to wave action and currents that are unique to aquatic environments. Also, reproduction of these weeds is due primarily to rapid vegetative growth, which results in clonal populations with little or no genetic diversity. Since many plant defenses against diseases and insects (including biocontrol agents) are determined by the genetic composition of a plant, the entire population of a clonally reproducing species would likely react to a biocontrol agent in the same manner; that is, if one plant is damaged by the biocontrol agent, the entire population is likely to be damaged by the agent as well. Waterhyacinth and the aquatic form of alligatorweed also are highly susceptible to secondary infection, so plants that have been injured by insects or disease rot and disintegrate very rapidly. Finally, beetles – especially weevils – have been responsible for most successful biocontrol programs. Adults of these insects tend to remain above the water, which may reduce fish predation, whereas larvae often feed inside the plant. These habits allow them to maintain high density populations in the environment. A number of successful weed biocontrol programs have utilized members of the insect group *Coleoptera*; in fact, the majority (greater than 75%) of insects released thus far for biocontrol of aquatic plants are weevils and beetles.

Defining success in biocontrol of weeds is usually subjective and highly variable. A project may be considered successful in an ecological sense when a biocontrol agent successfully establishes in an area and reduces the target weed's population. However, the severity of damage inflicted by the biocontrol agent may not result in the level of control desired by lake managers, boaters and homeowners. Recently, a clear distinction has been made between "biological success" and "impact success". Biocontrol agents can be biologically successful (they establish and sustain high population densities on the target weed), but may not realize impact success (they do not provide the desired level of control or impact on the weed).

The use of terms that define success (such as complete, substantial or negligible) in a biocontrol program may not take into account variations in time and space. For example, in the southeastern United States where the alligatorweed flea beetle has been introduced, biocontrol success can range from complete to negligible depending on the season, geographic area and habitat (Section 3.6.1). However, these terms can be useful from an operational perspective since they describe the current success level of biocontrol efforts and help managers to determine which other control measures (e.g., harvesters, aquatic herbicides) must be used to achieve the desired level of weed control. The advantage of this system is that it describes success in practical terms that are more readily understood by aquatic plant managers and the public. For example, biocontrol is defined as complete when no other control method is required, substantial when other methods such as herbicides are still required but at reduced levels and negligible when other control methods must be used at pre-biocontrol levels to manage the weed problem.

Summary

Biocontrol historically has been a major component of integrated pest management programs for terrestrial (crop, range, forestry, etc.) insect and weed control and can be an effective tool in the aquatic weed manager's arsenal as well. Classical biocontrol, which relies on importation of natural enemies from a weed's native home, may be useful to control an exotic invasive species that thrives when introduced to an area that lacks the natural enemies responsible for keeping the weed in check in its native range. The use of host-specific biocontrol agents allows management of populations of weedy species while leaving nontarget native plants unharmed. Successful biocontrol programs are often expensive and time-consuming to develop, but if successful can provide selective, long-term control of a target weed. Although a number of types of organisms – including disease-causing plant pathogens, insects and grass carp – have been studied for potential use as biocontrol agents for aquatic weeds, the greatest successes have been realized with insects (Section 3.6.1) and grass carp (3.6.2).

Photo and illustration credits:

Page 140: Biocontrol pipeline; Joshua Huey, University of Florida Center for Aquatic and Invasive Plants
Page 141: Biocontrol graph; Harley and Forno, 1992

3.6.1 Insects for Biocontrol of Aquatic Weeds

James P. Cuda: University of Florida, Gainesville FL; jcuda@ufl.edu

Introduction

Biocontrol of aquatic weeds with insects has resulted in the successful establishment of many potential biocontrol insects since it was first attempted in the US against alligatorweed in 1964. Aquatic weeds have historically been a more serious problem in the southern US due to the moderate climate and shallow lakes in these regions where weeds often cover large areas. Consequently, the greatest body of research on biocontrol has focused on weeds of the southern US. The following section describes in detail the relationship of particular biocontrol insects introduced into the US and their target weeds.

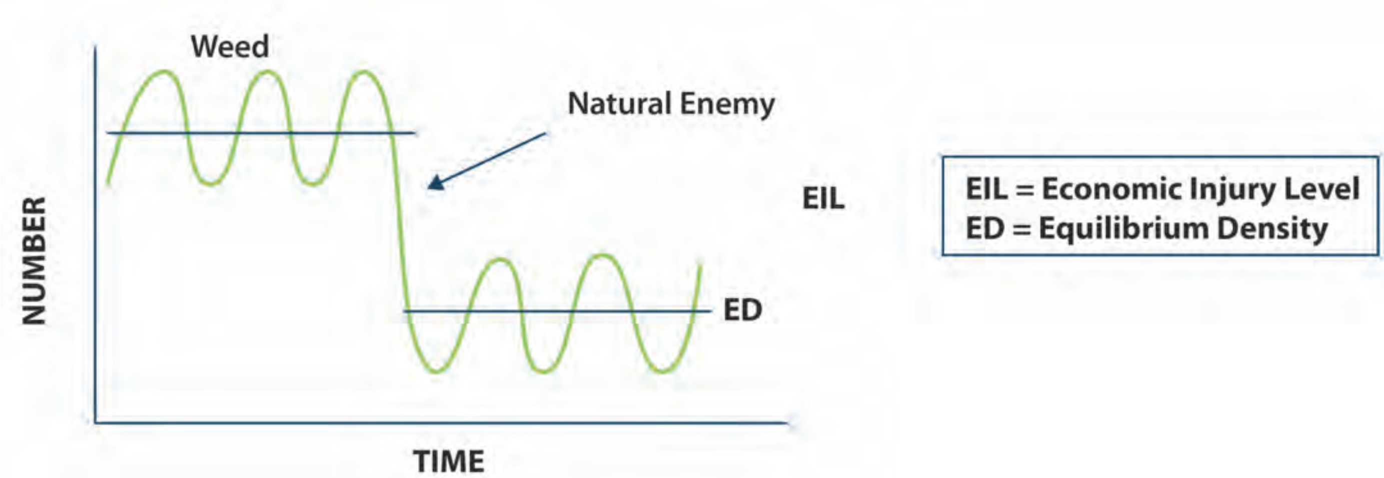
Alligatorweed (*Alternanthera philoxeroides*)

Enemy	Type	Origin (date)	Success	Comments
Agasicles hygrophila	Beetle	Argentina (1964)	Complete (south); negligible (north)	Found throughout the southern 2/3 of the range of alligatorweed in the US where it provides almost complete control
Amynothrips andersoni	Thrips	Argentina (1967)	Minimal	Attacks terrestrial alligatorweed more than the other species
Arcola (=Vogtia) malloi	Moth	Argentina (1971)	Moderate	Most important control agent in the upper Mississippi valley

If this manual had been written in the 1960s and 1970s, alligatorweed (introduced in the late 1800s) would have been included in Section 2 as one of the worst weeds in the US. The alligatorweed flea beetle (*Agasicles hygrophila*) was introduced in 1964 and has provided excellent control of the floating form of alligatorweed from southern Florida along the Gulf Coast to southern Texas. Unfortunately, the alligatorweed flea beetle is not as cold-tolerant as alligatorweed and insect populations die out during severe winters in the central and northern parts of the Gulf states. There are two growth forms of alligatorweed: plants that form floating mats along shallow shorelines and “terrestrial” plants that grow in intermittent wet/dry areas. The thrips feeds on terrestrial alligatorweed in northern areas especially in North Carolina. The beetle that is so effective in the southern range does not attack the terrestrial growth form.

Alligatorweed remains a problem in areas such as central and northern Texas, Mississippi, Alabama, Georgia and the Carolinas. The alligatorweed flea beetle is self-sustaining in its southern range but not in the north. The US Army Corps of Engineers periodically collects and re-releases the beetle in northern areas during spring to reestablish northern populations. This is an example of combining augmentation with classical biocontrol. The alligatorweed flea beetle has eliminated the need for other forms of control in natural areas when it is well-established. The *Amynothrips* and *Arcola* insects also are established on alligatorweed; the *Amynothrips* attacks the terrestrial alligatorweed plants more than do the other species. However, control of alligatorweed is largely attributed to the alligatorweed flea beetle. Alligatorweed provides a good example of how a biocontrol agent controls its weedy host plant without completely eradicating the population of the weed.

Alligatorweed grows quickly in spring and populations of the alligatorweed flea beetle increase as well, but lag behind development of the host plant. By the time alligatorweed has grown enough to become problematic, the population of the alligatorweed flea beetle reaches a density sufficient to destroy most of the alligatorweed. The number of alligatorweed flea beetles then decreases, alligatorweed growth resumes and this density-dependent cycle begins anew.



This is a nearly perfect example of a highly successful insect biocontrol program that adequately controls an invasive aquatic plant. Furthermore, 40 to 50 years after their introduction, none of the three insects released to control alligatorweed have been found feeding on, reproducing on or otherwise affecting nontarget native species.

Waterhyacinth (Section 2.11)

Enemy	Type	Origin (date)	Success	Comments
Neochetina bruchi	Weevil	Argentina (1974)	Substantial	Widely distributed throughout the range of waterhyacinth in the US
Neochetina eichhorniae	Weevil	Argentina (1972)	Substantial	Adults produce characteristic feeding scars on the leaves
Niphograpta albiguttalis	Moth	Argentina (1977)	Negligible	Prefers plants with short bulbous petioles
Orthogalumna terebrantis	Mite	USA (native)	Negligible	Produces characteristic dark stripes in the leaves; also attacks pickerelweed
Megamelus scutellaris	Bug	South America (2010)	Established	Damaged leaves and stems exhibit chlorosis

Two *Neochetina* weevils, the *Niphograpta* stem-boring caterpillar and the *Megamelus* planthopper have been released as biocontrol agents of waterhyacinth. The life cycle of the *Neochetina* weevils requires about 2 to 3 months to complete and is dependent on temperature. These weevils act on waterhyacinth by causing feeding damage that reduces the plant's ability to regenerate. Adult weevils produce characteristic rectangular feeding scars on the leaves, whereas larvae tunnel inside the leaf petioles to the crown or meristem where they damage new growth. Feeding damage also allows plant pathogens to invade the feeding scars and larval tunnels, which further weakens the plant. The life cycle of the *Niphograpta* caterpillar is completed in about 4 to 5 weeks. This insect prefers to attack smaller plants with bulbous petioles; petioles that are attacked often become waterlogged and die. However, the impact of the *Niphograpta* caterpillar has been difficult to evaluate because it causes tremendous damage for only a brief period and then disappears. The life cycle of the *Megamelus* planthopper from egg to adult takes about 25

days. Both the adults and nymphs cause extensive feeding damage that leads to chlorosis (yellowing) of the leaves and stems. Unlike the *Neochetina* weevils and *Niphograpta* caterpillar, *Megamelus* planthopper adults and nymphs feed externally and can disperse from waterhyacinth mats following chemical or mechanical control. The *Neochetina* weevils and the *Niphograpta* stem-boring caterpillar are established and occur almost everywhere waterhyacinth is distributed throughout the southern US. Growth of waterhyacinth is suppressed and vegetative reproduction is reduced, but other means of control are necessary in most areas. The *Megamelus* planthopper is currently at low densities due to weak dispersal behavior, interspecific competition with the weevils and egg parasitism.

Hydrilla (Section 2.2)

Enemy	Type	Origin (date)	Success	Comments
Bagous affinis	Weevil	India (1987)	Not established	
Bagous hydrillae	Weevil	Australia (1991)	Negligible	Adults recovered in 2009 from southern Louisiana See https://edis.ifas.ufl.edu/in1036
Cricotopus lebetis	Midge	Unknown (adventive)	Minimal?	Damages growing tips of hydrilla Also see http://edis.ifas.ufl.edu/IN211
Hydrellia balciunasi	Fly	Australia (1989)	Negligible	Found primarily in Texas See https://edis.ifas.ufl.edu/in1034
Hydrellia pakistanae	Fly	India (1987)	Minimal?	Widely distributed on dioecious hydrilla in the southeastern and south-central US
Parapoynx diminutalis	Moth	Asia (adventive)	Negligible	Causes localized heavy damage to hydrilla See https://edis.ifas.ufl.edu/in1024
Ctenopharyngodon idella	Fish	China (1963)	Substantial	Throughout the US by permit (Section 3.6.2) See http://plants.ifas.ufl.edu/guide/grasscarp.html

Two *Bagous* weevils (one from India that attacks tubers and one from Australia that mines stems) have been introduced as biocontrol agents for hydrilla. Only the Australian species has established but does not suppress hydrilla. The two introduced *Hydrellia* flies (one from India and one from Australia) have become established. The fly *H. pakistanae* is

widespread in the southern US, whereas *H. balciunasi* is localized in distribution. Populations of *Hydrellia* flies have not reached densities high enough to control hydrilla, possibly due to parasitism of the pupae by a native wasp or perhaps other environmental factors. The entire life cycle for both flies is completed in about 3 to 4 weeks, which should allow development of high insect populations. The adventive *Parapoynx* moth from Asia probably entered the US via the aquarium trade and was discovered in Florida feeding on hydrilla in 1976. The life cycle of *Parapoynx* is completed in 4 to 5 weeks. The moth was never studied or approved for release, but large populations of hydrilla are occasionally completely defoliated by the moth. The adventive naturalized nonnative *Cricotopus* midge has been associated with hydrilla declines in several Florida locations since 1992. The life cycle of *Cricotopus* is completed in 1 to 2 weeks and developing larvae of the midge mine the shoot tips of hydrilla, which severely injures or kills the plant's growing tips. Feeding damage changes the plant's structure or architecture by preventing new hydrilla stems from reaching the surface of the water column. Despite localized and occasionally severe impacts on hydrilla, none of these insects can cause damage significant enough to provide adequate control when used alone. Research to identify biocontrol agents for hydrilla continues due to the increasing spread of the species throughout the US, its development of resistance to the herbicides fluridone and endothall and the relatively high costs associated with other methods employed to control this weed.

Purple loosestrife (Section 2.18)

Enemy	Type	Origin (date)	Success	Comments
Galerucella calmariensis	Beetle	Germany (1992)	Substantial	Widely distributed throughout the range of purple loosestrife in the US
Galerucella pusilla	Beetle	Germany (1992)	Substantial	
Hylobius transversovittatus	Weevil	Germany (1992)	Substantial	
Nanophyes marmoratus	Weevil	France, Germany (1994)	Negligible?	

Two nearly identical *Galerucella* leaf beetles are responsible for most biocontrol of purple loosestrife; in fact, these beetles have reduced purple loosestrife infestations by 90% in several states, especially Oregon and Washington. Larvae feed on buds, leaves and stems of the plants and heavily defoliated plants are often killed by the feeding insects. The life cycle of the beetles is completed in about 6 weeks but there is only one generation per year, with pupation occurring

in the soil if it is not continuously flooded. This low rate of reproduction is responsible for the lag time between introduction of the beetles and noticeable effects on the plants. Two weevils – the root-attacking *Hylobius* shown here and seed-attacking *Nanophyes* – also contribute to the successful biocontrol of purple loosestrife. Larvae of *Hylobius* feed and develop in the tap roots and pupation occurs in the upper part of the root. Larvae require 1 to 2 years to complete their development and adults can live for several years. Adults of *Nanophyes* feed on young leaves or flowers and lay their eggs in flower buds. Pupation occurs inside the bud and larvae consume the flower buds; buds then fail to open and drop prematurely from the plant. Although the entire life cycle is completed in about 1 month, there is only 1 generation per year. Leaf-eating *Galerucella* beetles, root-attacking *Hylobius* weevils and seed-attacking *Nanophyes* weevils have only recently been introduced as biocontrol agents on purple loosestrife but appear to be very successful in reducing the growth, occurrence and competitiveness of this emergent weed.

Eurasian watermilfoil (Section 2.3)

Enemy	Type	Origin (date)	Success	Comments
Acentria ephemerella	Moth	Europe (adventive)	Negligible?	All can cause declines to populations of Eurasian watermilfoil in localized areas of lakes. Results are difficult to predict.
Cricotopus myriophylli	Midge	China (adventive)	Negligible?	
Eurychiopsis lecontei	Weevil	US (native)	Substantial?	

Several insects have been found attacking Eurasian watermilfoil during overseas surveys, but none have been introduced to the US thus far. Recent declines in the abundance of Eurasian watermilfoil in some northern lakes have been attributed to the adventive *Acentria* moth and *Cricotopus* midge, as well as the native *Eurychiopsis* weevil. These insects are widely distributed throughout the range of Eurasian watermilfoil in North America and are found in all areas infested by the weed; as a result, it is difficult to assess their effectiveness as biocontrol agents. Larvae of the *Acentria* moth feed both in and on stems and leaves, which causes the leaves to drop off the plant. Females have reduced wings and are usually flightless and mating occurs in or on the water surface. Two generations are produced annually and pupae form on the stems. Larvae also feed on a variety of native plants in the absence of Eurasian watermilfoil, so the *Acentria* moth is not a typical biocontrol agent. The *Cricotopus* midge is widely distributed and has been shown to reduce the growth and biomass of Eurasian watermilfoil in laboratory experiments. This midge is not the same species of *Cricotopus* that attacks hydrilla, which suggests these insects may be host specific. It is worth noting that midges rarely feed on living plant tissue and most species typically feed on decaying organic matter. The *Eurychiopsis* weevil is generally considered to be the most important biocontrol agent of Eurasian watermilfoil from an operational perspective. Even though it is a native insect, this weevil prefers Eurasian watermilfoil over its native natural host. The life cycle of the weevil is completed in about 30 days; adults feed on leaves and stems, whereas larvae are stem borers that consume apical meristems. Feeding damage causes the stems to break apart and heavy feeding by the insects prevents the formation of surface mats. High populations of the *Eurychiopsis* weevil have been associated with declines of populations of Eurasian watermilfoil in some northeastern and midwestern states but fish predation may prevent this weevil from reaching its full biocontrol potential. The *Eurychiopsis* weevil is commercially available and can be purchased to augment existing weevil populations. However, research studying the value of augmenting existing populations with purchased insects has been inconclusive.

Waterlettuce (Section 2.12)

Enemy	Type	Origin (date)	Success	Comments
Spodoptera pectinicornis	Moth	Thailand (1990)	Not established	May be affected by predation by other insects
Neohydronomus affinis	Weevil	Brazil (1987)	Negligible?	

Waterlettuce is a tropical species that is believed to be native to North America and was extirpated (died out) during the Ice Ages but was reintroduced into Florida in the 16th century. It forms large floating mats similar to those of waterhyacinth in the extreme southern US and populations of waterlettuce often increase as waterhyacinth populations decline. Waterlettuce is a public health issue in Florida, where larvae of disease-causing *Mansonia* mosquitoes (Section 1.5) attach to the extensive feathery roots to obtain oxygen. Two insects have been released as biocontrol agents of waterlettuce but only the *Neohydronomus* weevil has become established.

Adults and larvae of the *Neohydronomus* weevil feed on the leaves, crown and newly emerging shoots of waterlettuce and the characteristic "shot hole" appearance of leaves indicates high weevil densities. Feeding by multiple larvae destroys the spongy leaf bases, which causes plants to lose buoyancy. The life cycle of the *Neohydronomus* weevil is completed in 3 to 4 weeks. The weevil has not contributed to long-term suppression of the plant in the US but has provided successful biocontrol of waterlettuce in other countries. It is thought that the *Neohydronomus* weevil is heavily preyed upon by imported fire ants in Florida; if true, this provides an interesting example of an exotic invader controlling a valuable potential biocontrol agent.

Giant salvinia (Section 2.13)

Enemy	Type	Origin (date)	Success	Comments
Cyrtobagous salviniae	Weevil	Brazil? (adventive)	Negligible, substantial	Provides good control of common salvinia in Florida and Louisiana. Effects of 2001 introduction on giant salvinia are dramatic at some sites
Cyrtobagous salviniae	Weevil	Brazil (2001)	Substantial	

The *Cyrtobagous* weevil is the only insect that has been released as a biocontrol agent of giant salvinia. Adventive weevils that were discovered in Florida in 1960 control common salvinia (*Salvinia minima*), whereas weevils released in 2001 from a Brazilian population are used as biocontrol agents for giant salvinia. The entire life cycle of the *Cyrtobagous* weevil takes about 46 days. Adults feed on leaf buds and leaves and larvae tunnel inside the plant, killing leaves and rhizomes. Attacked plants turn brown and eventually lose buoyancy. *Cyrtobagous* weevils are currently of great interest to aquatic plant managers worldwide due to their ability to effectively control giant salvinia.

Melaleuca (*Melaleuca quinquenerva*)

Enemy	Type	Origin (date)	Success	Comments
Oxyops vitiosa	Weevil	Australia (1997)	Substantial	Not established in permanently flooded sites due to inability to complete life cycle. See http://edis.ifas.ufl.edu/document_in172
Boreioglycaspis melaleucae	Psyllid	Australia (2002)	Substantial	
Fergusonina turneri	Fly	Australia (2005)	Not established	
Lophodiplosis trifida	Fly	Australia (2008)	Substantial	Feeding damage can kill melaleuca seedlings and saplings. See https://edis.ifas.ufl.edu/in1140

Melaleuca is a locally invasive plant that occurs only in south Florida and the Everglades and was introduced multiple times during the early 1900s. The species was used as an ornamental tree and was planted in marshes to drain wetlands. Melaleuca typically grows in dense, impenetrable stands and can attain a height over 50 feet. Four insects have been released as biocontrol agents of melaleuca but only three have become established.

The *Oxyops* weevil and the *Boreioglycaspis* psyllid were released in 1997 and 2002, respectively, and are widely established on melaleuca in south Florida. Damage to the tree is caused primarily by the immature stages of these insects. The slug-like weevil larvae feed on newly expanding leaves; psyllid nymphs attack older leaves and woody stems in addition to new leaves and the psyllid can kill newly emerged seedlings as well. These two insects complement each other well; the psyllid is able to complete its development entirely in the tree canopy under flooded conditions that prevent establishment of the weevil, which must pupate in the soil. Extensive leaf damage from both insects causes melaleuca to divert resources to the production of new foliage instead of flowers. The life cycle of the weevil is completed in about 3 months, whereas a new psyllid generation is produced in 6 weeks. The *Oxyops* weevil and the *Boreioglycaspis* psyllid have contributed to the substantial biocontrol of melaleuca. The *Lophodiplosis* gall-forming fly was released in 2008. It takes about six weeks from the time that the larva emerges from the egg until the midge becomes an adult. Gall formation diverts the tree’s resources from normal growth and reproduction, and enhances the effects of other biological, chemical, and mechanical controls.

Summary

The use of insects as biological control agents for aquatic weeds has yielded mixed results, which is typical and expected of biocontrol programs. A number of aquatic weeds – including alligatorweed, purple loosestrife, melaleuca and salvinia – are being successfully controlled by insects released as biocontrol agents for these species. Control of other aquatic weeds – including waterhyacinth, hydrilla, Eurasian watermilfoil and waterlettuce – has been less successful. Multiple factors play a role in the failure of some biocontrol agents to reach their full potential. For example, the *Neohydronomus* weevil has provided successful biocontrol of waterlettuce in other countries, but has failed to control waterlettuce in Florida, possibly due to predation of the weevil by imported fire ants. Biocontrol can be an effective tool in the aquatic weed manager’s arsenal since host-specific biocontrol agents allow management of populations of weedy species while leaving nontarget native plants unharmed. Therefore, it is important that researchers continue to identify and evaluate biocontrol agents so that the successes realized in the control of alligatorweed, purple loosestrife, salvinia and melaleuca can be duplicated in other weedy aquatic species. A major factor that limits the utility of biocontrol is that unless a potential biocontrol agent is species-specific, it cannot be introduced into the US. Therefore, it is unlikely that biocontrol alone can control all the invasive aquatic weeds in the US.

Photo and illustration credits:

Page 145: Alligatorweed flea beetle; Gary Buckingham, USDA-ARS
Page 146 upper: Graph; from Harley and Forno, 1992
Page 146 lower: *Neochetina bruchi* weevil; CSIRO, distributed under a CC BY 3.0 license
Page 147 upper: *Megamelus scutellaris*; Philip Tipping, USDA ARS IPRL
Page 147 lower: *Hydrilla verticillata*; William Haller, University of Florida
Page 148: Adult root-boring weevil *Hylobius transversovittatus*; Bernd Blossey
Page 149: *Cyrtobagous salviniae* on giant salvinia frond; Scott Bauer, bugwood.org

3.6.2 Grass Carp for Biocontrol of Aquatic Weeds

Mark Hoyer: University of Florida, Gainesville FL; mvhoyer@ufl.edu

The grass carp (*Ctenopharyngodon idella* Cuvier and Valenciennes), is native to rivers in eastern Russia and China that flow into the Pacific Ocean, but the fish has been introduced to approximately 70 countries including the US, Taiwan, Japan, Mexico, India, Malaysia and several European countries. It has been introduced for use as both a food fish and for biological control of aquatic weeds. Although the grass carp is highly adaptable and can survive in a variety of

conditions, the natural grass carp life cycle has not been observed to occur many times outside of the native range. The restriction is related to reproduction since the fish requires large and long low gradient, turbid rivers and cannot reproduce in confined water bodies. Of all the countries where the fish were introduced, they have established populations in only few countries, primarily in Asia and Europe. However, there have been reports of breeding populations including in the Atchafalaya, Mississippi (and major tributaries) and the Trinity rivers in the US.

Grass carp were imported to the US in 1963 as a possible biological control agent for hydrilla (Section 2.2) and other aquatic plants. Their arrival caused an explosion of research on their use as an aquatic plant management tool. Efficacy experiments were conducted in Florida in the 1970s by the United States Department of Agriculture and the University of Florida. Use of the "diploid" fish was limited from 1970 until 1984 due to tight regulations developed to address their possible escape and reproduction, which could potentially impact native flora and fauna. These concerns led to research that developed a sterile (non-reproductive) triploid fish, which was equally effective in controlling hydrilla. Sterile fish were developed by subjecting fertilized eggs to stress, such as temperature (hot or cold) or pressure. The stress causes each egg to retain an extra set of chromosomes and become triploid instead of diploid. Although triploid fish are sterile, this does not affect their aquatic plant herbivory.

Grass carp are so effective at controlling aquatic plants that in 2009 the use of grass carp was recorded in 45 states, the exceptions being Alaska, Maine, Montana, Rhode Island and Vermont. Most states currently require that only artificially produced sterile triploid grass carp be stocked to prevent further natural reproduction in our large river systems. The use of grass carp for aquatic weed control is governed by individual states; some require permits, site inspections and use of sterile fish, whereas others have no restrictions. Several states in the northern US actually prohibit the possession, sale or transportation of grass carp. As a result, you must consult the appropriate state agencies before considering grass carp for weed control to determine whether their use is restricted or prohibited in your state

Consumption rates and aquatic plant preferences

Grass carp consumption rates (measured as the daily percentage of body weight eaten) are affected by size of the fish and by environmental characteristics such as temperature, salinity and oxygen content of the water. Also, grass carp consumption rates decrease as fish become larger and reach sexual maturity (which occurs even in sterile fish) at 2 or 3 years of age. Large grass carp (over 15 pounds) consume up to 30% of their body weight daily, whereas smaller fish (less than 10 pounds) can consume as much as 150% of their body weight each day. Maximum consumption occurs when water temperatures range from 78 and 90 °F and is greatly reduced when temperatures are below 55 °F. Consumption is reduced by 45% when oxygen levels in the water drop to 4 ppm and fish stop feeding completely if the oxygen level drops below 2 ppm. Although grass carp can tolerate salinities up to 10 parts per thousand, they will not feed if salinity levels are higher than 6 parts per thousand.

The grass carp is a general herbivore and feeds on vegetation mostly near the surface of the water. Many plant palatability studies have been conducted to determine grass carp plant preferences. Plant preference is determined primarily by the aquatic plant's structural firmness as well as the secondary metabolites produced by a particular plant species. However, the grass carp is a generalist, and in the absence of the preferred plant, will feed on most other types of aquatic vegetation. Grass carp even have been observed to feed on terrestrial plants that are hanging over the water. In general, the five most-preferred species in order of preference are hydrilla, musk grass (*Chara* spp.), pondweeds (*Potamogeton* spp.), southern naiad (*Najas guadalupensis*) and egeria (Section 2.5). Grass carp rarely provides good control of filamentous algae (Section 2.18), Eurasian watermilfoil (Section 2.3), spatterdock (*Nuphar advena*), fragrant waterlily (*Nymphaea odorata*), sedge (*Cladium* sp.), cattail (*Typha* sp.) or other firm-structured aquatic plants.

Stocking rates and duration of aquatic plant control

Symposia held during 1979 and 1994 (Grass Carp Symposia), 2004 (Hydrilla Management in Florida) and 2008 (Triploid Grass Carp Risk Analysis) summarized hydrilla management strategies as well as the potential of using grass carp as a management tool. Additional studies that evaluated methodologies, impacts or controversies associated with grass carp were conducted in Lake Conroe TX, Lake Guntersville AL and the Santee Cooper reservoirs in South Carolina. Most of these studies determined that introducing triploid grass carp is the most cost-effective method for long-term control of hydrilla but with a major limitation: triploid grass carp are best used where the loss of all palatable submersed vegetation is an acceptable outcome for an extended period of time.

Grass carp should not be stocked in open systems that are connected to a stream or river because they migrate with moving water and will leave the stocked water body, thus not providing any plant control. Grass carp stocking rates in closed systems typically range from 2 to 50 fish per acre. Most biologists agree that there is no "magic stocking number" of grass carp to achieve a specific percentage of submersed weed control. Both growth rates of aquatic plants and consumption rates of grass carp are moving targets that are dependent on many factors including but not limited to the type and quantity of aquatic plants present, water clarity, oxygen content and nutrient concentration of the water. If the consumption rate of the grass carp exceeds the growth rate of the aquatic plants, then plant control can be achieved; otherwise, plant abundance generally remains a problem.

Nationally, because grass carp feeding rates are reduced in cooler water, stocking rates in northern temperate climates may need to be higher than in southern subtropical climates to achieve similar plant control. The mortality rate of grass carp immediately after stocking varies tremendously and can impact the success or failure of the plant management action. Once grass carp are stocked, predation by fish-eating predators can be a problem because grass carp typically feed near the water surface and are commonly targeted by osprey, otters and other fish. For example, studies in research ponds in Florida revealed that the number of grass carp lost to predation ranged from 7 to 70% one year after stocking. Predation can be especially problematic in water bodies with large fish predators such as striped bass or largemouth bass. Grass carp that are larger than 12 inches should be used in these systems to avoid losing the majority of the stocked grass carp to predation and to ensure adequate aquatic weed control. Overstocking or excellent survival of stocked grass carp often results in the depletion of almost all submersed aquatic plants, whereas understocking or excessive mortality of grass carp results in no noticeable plant control. The proper balance of grass carp and weed growth is difficult/impossible to achieve and only a few cases of this type of success have been reported.

If complete elimination of aquatic plants by grass carp is an outcome, it can last for as long as 10 to 15 years. It is important to understand that the use of grass carp as biocontrol agents is a long-term strategy because grass carp grow to an extremely large size, live up to 20 years and cannot easily be removed from a water body once they are stocked. It is almost impossible to remove significant numbers of grass carp (approximately 90% of population) from large lakes to allow the growth rate of aquatic plants to exceed the consumption rate of the carp, thus allowing an increase in aquatic plant abundance. Many studies have examined dozens of grass carp removal methods from lakes and none were efficient enough to remove a significant number from a lake and/or canal. However, a few management agencies are still experimenting with removal techniques and in some cases bowfishing tournaments show promise as a means of removing grass carp from lakes (see https://fishgame.com/2016/10/bowfishing-grass-carp-solving-overpopulation-problem/). Additionally, it may take five to 20 grass carp per acre to achieve aquatic plant control but it only takes approximately one carp per acre to maintain control.

Effects on water quality and fish populations

Most changes in water quality and plankton abundance associated with grass carp are not caused directly by the fish but are due to the removal of aquatic plants and associated periphyton (algae attached to the plant material). When aquatic plants are controlled in a lake with grass carp, herbicides (Section 3.7.1) and/or mechanical cutting (Section 3.5), all of which leave dead plant material in the lake, nutrient and algal abundance tends to increase and water clarity tends to decrease (Section 1.1). These whole-lake changes in water quality only occur when large amounts of plants (greater than 15 to 30% coverage of aquatic plants) are controlled and the changes are related to three primary mechanisms: 1) by ingesting plant material and associated periphyton, nutrients incorporated into plant tissue are released, which increases ambient nutrient concentrations, causes more algal growth and fills the water column with algal cells while decreasing water clarity as measured by Secchi depth readings, 2) aquatic plants also tend to stabilize the water column not allowing wind to re-suspend sediments and nutrients, and 3) the stabilized water allows the sedimentation of algal cells and other detritus, which results in a low-nutrient and clear-water state.

Similar to water quality, changes in fish population characteristics associated with grass carp are not caused directly by the fish, but are due to the removal of aquatic plants. Canfield and Hoyer (1992) examined fish populations in 60 Florida lakes that varied in lake trophic status and abundance of aquatic macrophytes with the goal of determining the optimum abundance of aquatic macrophytes for a healthy fish population. They reported that fish populations functioned normally in lakes with aquatic plant coverage of 15 to 85% and that fish populations in lakes with less than 15% or greater than 85% plant coverage were more likely to be depressed. Therefore, if grass carp reduce aquatic plant abundance to zero, there is an increased likelihood of having a depressed fish population. However, there are many emergent and floating-leaved aquatic plants that grass carp cannot or will not consume, so most lakes with grass carp always maintains some habitat that is sufficient for some sportfish, especially in smaller lakes (approximately 500 acres or less).

Summary

Grass carp can be an effective, cost-efficient tool for long-term aquatic plant removal and management in closed systems. However, management agencies and stakeholders that use the system targeted for stocking with grass carp must be willing to accept the probability that all submersed aquatic vegetation may be eliminated and with the elimination of the submersed aquatic vegetation will also come changes in water quality and fish populations. Additionally, until new removal techniques are developed the users must accept that there is no way to affectively remove the carp once they are stocked.

Photo and illustration credits:

Page 151: Mature grass carp; William Haller, University of Florida
Page 152: Releasing grass carp; William Haller, University of Florida
Page 153: Grass carp; Paul Shafland, Florida Fish and Wildlife Conservation Commission

3.7 Requirements for Registration of Aquatic Herbicides

Don Stubbs: US Environmental Protection Agency (retired); donald271@verizon.net
Carlton R. Layne: Environmental Protection Agency (retired) and Executive Director, Aquatic Ecosystem Restoration Foundation; clayne@aquatics.org

This section discusses the changes in pesticide regulation in the US over the past 50 years when the new US Environmental Protection Agency (EPA) developed an emphasis on evaluating the registration and use of pesticides based on research on human health and environmental concerns.

History of pesticide regulation

A pesticide is defined as any product that claims to control, kill or change the behavior of a pest. The United States first started regulating pesticides in 1910. The 1910 Federal Insecticide Act was intended to protect farmers from adulterated products and false labeling claims. With the continuous increase in pesticide development and use after World War II, Congress passed the Federal Insecticide, Fungicide and Rodenticide Act (FIFRA) in 1947. This act, which would be amended through the years, required that all pesticides be registered with the Department of Agriculture before they could be shipped in interstate commerce. The same federal agency responsible for agricultural production in the United States was now responsible for the regulation of pesticides on agricultural crops. FIFRA established procedures for the registration and labeling of pesticides but dealt mainly with the efficacy or effectiveness of pesticides and did not regulate pesticide use. Almost anyone could use a pesticide for any purpose and there was no legal recourse if a pesticide was not properly used. In addition, FIFRA did not allow for the denial of a pesticide registration request.

In 1962 Rachel Carson published “Silent Spring”, which drew widespread public attention to the indiscriminate use of pesticides with unknown human health and environmental effects. Many of the pesticides were persistent in the environment and were transferred from one animal to the next upon being eaten (a phenomenon known as bioaccumulation). As a result, some pesticides were ultimately ingested by humans and other nontarget animals, including wildlife. Very little was known at the time about the fate of pesticides in the environment and the potential effects of their residues on man and wildlife.

The Environmental Protection Agency (EPA) was created in 1970 and the responsibility for regulating pesticide use and labeling was transferred from the USDA to this new agency. This marked the beginning of a shift in the focus of federal policy from the control of pesticides for reasonably safe use in agricultural production to the control of pesticides for the reduction of unreasonable risks to man and the environment. In 1972 Congress passed the Federal Environmental Pesticide Control Act, which amended FIFRA and set up the basic American system of pesticide regulation to protect applicators, consumers and the environment that we have today. This Act gave the EPA greater authority over pesticide manufacturing, distribution, shipment, registration and use. EPA could now, among other things:

1) require additional data as necessary;
2) suspend or cancel the registration of existing pesticides;
3) prohibit the use of any registered pesticide in a manner inconsistent with label instructions;
4) require that pesticides be classified for specific uses;
5) deny a registration request;
6) provide penalties (fines and jail terms) for violations of FIFRA;
7) provide states with the authority to regulate the sale or use of any federally registered pesticides in that state as long as state rules were at least as strict as federal guidelines.

In 1988 Congress once again amended FIFRA by requiring the EPA to reregister all pesticides registered before November 1984 and to ensure that the database was current and in accordance with modern science. The development of the Food Quality Protection Act (FQPA) in 1996 amended both the FIFRA and the Federal Food, Drug, and Cosmetic Act (FFDCA). This Act set a single health-based standard for residues of pesticides in food and required the EPA to reevaluate all tolerances for pesticides and their inert ingredients.

Registration

Pesticide regulations are continuously under review and revision as scientific methods and knowledge increase. The following parts of this section will discuss pesticide registration and enforcement of pesticide laws, which are just a portion of the EPA's overall responsibility to protect the environment. It costs $30 to $60 million or more, and 8 to 10 years, to introduce a new pesticide to the market. Pesticides that are destined for use in aquatic systems in the US must be registered by the federal government through the EPA and by the state in which the pesticide will be used. The product may only be used in accordance with the label accepted by the EPA and any other applicable state regulations as long as the state regulations are at least as restrictive as the federal label. A pesticide may occasionally be registered by a state based on a special local need. In such circumstances, the active ingredient of the pesticide must be registered by the EPA and the appropriate tolerances in fish, shellfish and irrigated crops must be established by the EPA. This federal agency has overall responsibility for pesticide regulation even in states with small but locally important pest control needs.

The burden of proof to show that a pesticide will not cause unreasonable adverse effects on man and the environment rests with the registrant (the company that develops or labels the pesticide). The registrant is responsible for testing the active ingredient and the end use product (the final formulated product offered for sale) for potential harm to man and the environment. The EPA requires between 84 and 124 different studies to satisfy this requirement. These studies include toxicity and exposure tests on laboratory animals that measure the possible effects of the pesticide on human

health – to applicators and to the general public – through direct exposure and through residues in food. These studies also determine the fate of the pesticide once it is introduced into the environment and the effect of the pesticide on nontarget organisms. The EPA reviews these studies and determines the appropriate labeling for the use of each pesticide. Label precautions may include user safety information (protective clothing, reentry intervals or specific hazards), environmental safety warnings, container disposal and pesticide classification. In addition, all labels must provide appropriate directions for use (see "Pesticide Labeling" below).

The EPA regulates pesticide use from occupational (applicator/worker), residential and dietary standpoints and determines the potential effects of acute (immediate or short term), intermediate and chronic (long term) exposure to humans. If the use of a pesticide results in a residue of the pesticide in food or feed, it is necessary to establish a tolerance (maximum legal residue) level for that pesticide under the FFDCA. The EPA also evaluates residues in drinking water and must determine whether pesticide residue levels found in drinking water, fish, shellfish and any other food or feedstock meet the safety standard of the FQPA. In short, the EPA verifies that there is a reasonable certainty that no harm will result from the residues of the pesticide in food or feed. The FQPA is a risk-based statute and does not provide for the analysis of risks vs. benefits. Examples of some of the studies required before a product can be used as a pesticide are listed below. More detailed information is available at https://www.epa.gov/pesticide-registration/about-pesticide-registration.

Toxicity studies (how dangerous is the pesticide to humans?)

•Acute toxicity: study the immediate/short term effects of exposure to determine appropriate user precautions
•Sub-chronic toxicity: examine intermediate toxicological effects to identify the risks of less than lifetime exposure
•Chronic toxicity: evaluate long-term toxicity effects to determine possible problems associated with a lifetime of exposure
•Oncogenicity: determine whether the product causes cancer
•Developmental and reproductive toxicity: identify any effects on development and reproductive function

Chemistry studies (what is the pesticide?)

•Chemical identity, physical and chemical properties
•Disclosure of manufacturing process and all inert ingredients
•Determine chemicals of concern including the active pesticide and inert components
•Develop analytical methods for determining concentrations of the pesticide in plants, soil, water and food
•Determine the amount of pesticide left on plants, soil, water and food as a result of use

Environmental fate (what happens to the pesticide after it has been applied?)

•Hydrolysis: establish the significance of chemical breakdown in water
•Photolysis: determine the interaction of the pesticide under natural light
•Degradation: determine when the pesticide breaks down and what it breaks down to (metabolites) in water, soil and air
•Metabolism: examine the breakdown of the pesticide by organisms in the soil and water under both aerobic and anaerobic conditions
•Mobility and bioaccumulation: determine how the pesticide moves in the environment and whether it accumulates up the food chain
•Field dissipation: test and monitor how the pesticide and any major metabolites behave under realistic conditions

Ecological toxicity (how dangerous is the pesticide to fish, birds, mammals, invertebrates/pollinators, and plants?)

•Acute toxicity: study the immediate effects on wildlife
•Chronic dietary toxicity: examine the effects of a lifetime of exposure in birds
•Reproduction studies
•Toxicity to plants

Because the EPA relies on data submitted by the registrant, it carries out a laboratory audit program. This program sends EPA scientists and enforcement personnel to laboratories that conduct studies on pesticides. These personnel are responsible for reviewing the testing procedures to ensure that they are carried out in accordance with EPA regulations for conducting accurate laboratory studies. These conditions, known are Good Laboratory Practices, include strict guidelines on how studies are conducted and documented. In addition, the EPA requires that the registrant submit to them any data concerning adverse effects associated with the use or new testing of the chemical. These data are immediately reviewed by the EPA and any corrective action (label changes, use deletions or product cancellation) is taken as deemed necessary by the agency.

Tolerances

A tolerance is a residue level established by regulation which is considered a “safe level” of a pesticide and it is also an enforceable level. An “enforceable level” essentially means that when a pesticide is found in or on a food product and is either (1) not registered for use on that food product, or (2) present at a level higher than the tolerance established for that food crop, the food crop may be destroyed and investigations must be conducted to determine whether fines or other penalties are warranted. The tolerance is based on acute and chronic animal toxicity data. These data are multiplied by a 100-fold safety factor to determine an allowable residue level. The EPA does not set tolerances in drinking water as a result of pesticide use, but it does assess the safety of drinking water using the same safety standard for water as it does for food or feed before it will register the pesticide. Under the FFDCA as amended by the FQPA in 1996, a tolerance may only be established when the EPA determines that there is a reasonable certainty that no harm will result from the aggregate exposure (food, water and residential exposure) to the active ingredient and the inert ingredients in the pesticide.

Pesticides that are registered for use in a way that results in residues of the pesticide or its metabolites of concern in or on food or feed require the establishment of a tolerance under the FFDCA. Tolerances for pesticides are established under the FFDCA by the EPA. Food or feed contaminated with residues of pesticides or their metabolites of concern that do not have an established tolerance or have residues above the established tolerance level are considered adulterated and may be seized and destroyed by the Food and Drug Administration (FDA). While the EPA sets these pesticide tolerances, the FDA is responsible for enforcing them. Pesticides to be used in aquatic systems must have established tolerance levels of that pesticide and its metabolites of concern in fish, shellfish and any crops that would be irrigated with treated water.

Pesticide labeling

Pesticides are classified as either “general use”, which can be purchased and used by anyone, or “restricted use”, which may only be sold to and used by persons under the direct supervision of a certified applicator. A certified applicator must complete the appropriate federal or state training and testing. Pesticides can be used to control nuisance aquatic weeds without causing unreasonable adverse effects to man or the environment as long as label directions, precautions and warnings are followed.

The EPA regulates pesticides through pesticide labeling and determines the appropriate minimal label information required for the safe and effective use of the pesticide based on data submitted by the registrant. All labels must also include certain information; for example, all labels must carry several specific statements, including “Keep Out of Reach of Children” and a signal word (Caution, Warning or Danger). Directions for use – including application rates, number of applications allowed per season, user precautions, environmental precautions, container disposal instructions and other directions as determined by the EPA – are also required. In addition, every label must carry the statement “It is a violation of Federal law to use this product in a manner inconsistent with its labeling”. This means the pesticide can only be used in accordance with the label on the product container. The EPA stamps the label as accepted and this is the only label the registrant may place on its pesticide container before selling the product to the public. This label then becomes the principal communication between the registrant and the user. The directions for use, precautions and warnings tell the user how to use the pesticide and what precautions to take when the pesticide is used. Any changes to the labeling must be submitted to and approved by the EPA prior to marketing. For a full discussion on labeling requirements, please visit the EPA website on labeling at https://www.epa.gov/pesticide-registration/label-review-manual.

Review of registered pesticides

In 2008 the EPA completed its reregistration of all pesticides registered prior to November 1984 as required by the 1988 amendment to FIFRA. This effort took over 20 years as it required the reassessment of all products and their associated tolerances. In 2008 the EPA also initiated a Registration Review Program. This program, required by the 1996 amendments to FIFRA (FQPA), will review the registration of all registered pesticides on a continual 15-year cycle to ensure that pesticides remain in compliance with developing changes in science, public policy and pesticide use practices.

Good laboratory practices (GLP)

Working closely with the Office of Pesticides Programs, teams of investigators and scientists regularly conduct Good Laboratory Practices inspections at facilities that generate the scientific studies used in support of pesticide registrations. In addition, specific studies are randomly audited to verify adherence to identified protocols and procedures. Everything from the credentials of the researchers to the calibration of the equipment is thoroughly examined. The raw data are compared to the reported results to ensure accurate reporting. "For cause" audits of data are conducted when EPA scientists observe inconsistencies or irregularities in the studies submitted by the registrants.

FIFRA enforcement

General

Without enforcement, statutes and regulations are simply suggestions. A fair and vigorous enforcement program levels the playing field for the regulated community, removes any economic advantage of noncompliance (such as when using an unregistered pesticide on a site or crop not listed on the label) and exacts retribution as appropriate. As a result, enforcement is the exclamation point of the process that began with the registration of pesticides and the development of the labels and completes the mission of the EPA to provide a measure of consumer protection and to protect human health and the environment.

To ensure compliance with the requirements of FIFRA, federal agents and state inspectors monitor the marketplace and conduct inspections and investigations at establishments where pesticides are produced and distributed and at the facilities of commercial and private applicators where pesticides are stored. While all enforcement efforts are important, use-related inspections and investigations provide ongoing feedback to the EPA regarding the effectiveness of label requirements and accepted directions for use. This information, coupled with the requirement that registrants report all unanticipated adverse effects encountered as part of the distribution, sale and use of a pesticide, provides an impetus for additional data requirements. Mandatory label modifications may also be ordered depending on the nature of the data received.

Misuse

It is a violation of federal law for any person to use any registered pesticide in a manner inconsistent with label directions. The directions can cover all aspects of the pesticide's use, including transportation, storage, mixing, loading, application rates, target pests, use sites or crops, methods of application, personal and worker protection, environmental warnings, disposal and anything else necessary to protect human health or the environment. Federal and state inspectors conduct both routine facility inspections and "for cause" use investigations. Evidence of misuse (e.g., samples, photos, statements and records) may be used to prosecute violators in federal or state jurisdictions (or in both) depending on the circumstances of the case. Penalties can be substantial. For example, FIFRA provides for a $7,500 civil/administrative fine for each violation or count. In addition, criminal prosecutions are not unusual. While classified as misdemeanors, criminal offenses under FIFRA are considered serious environmental crimes and carry a maximum penalty of one year in jail per count plus substantial fines. Years ago, two unlicensed pest control operators in Mississippi were sentenced to 5.5 and 6.5 years in a federal penitentiary, respectively. Sentences of 2 to 3 years plus fines for the criminal misuse of pesticides are commonplace.

Product claims

As aquatic-related issues such as harmful algae blooms, unusual invasive species, and unusual environmental circumstances continue to appear, more and more creative responses to these issues arise that may require some sort of response by enforcement agencies. The EPA has tried to address some of these this in their Label Review Manual:

> For certain aquatic use products, claims to reduce sludge and unpleasant odors in water or to clean, clarify or deodorize ponds and lakes are not considered pesticidal claims; nor are claims regarding the reduction of nutrients and organic matter in water, **provided no claim is directly made or implied that the reductions will result in reduced pest populations** [emphasis added]. The claims "Reduces critical nutrients for cleaner, clearer ponds", "Ponds with algae need to reduce nutrients", and "Bacterial Product to Control Excess Nutrients for Clear, Clean Ponds" **imply pesticidal use and therefore require registration** [emphasis added].

Slime and odor control agents and other products expressly claiming control of microorganisms of economic or aesthetic significance are not considered to be public health-related but should bear accurate pesticide labeling claims. Registrants are still responsible for ensuring that these products perform as intended by developing efficacy data, which must be kept on file by the registrant.

EPA's policy does not permit the use of the terms "natural", or "naturally" in the labeling of any registered product, including biopesticide products, both microbials and biochemicals. These terms cannot be well defined and may possibly be misconstrued by consumers as a safety claim.

Aquatic dyes intended to reduce UV light or to otherwise reduce or control aquatic plants, algae or cyanobacteria are considered to be pesticides and must be registered with the EPA prior to distribution and sale.

Pesticide devices

Another area of increasing concern for EPA and state enforcement offices involves pesticide devices. In general, if an article is an instrument or contrivance that uses physical or mechanical means to trap, destroy, repel, or mitigate any plant or animal life, it is considered to be a device and is subject to regulation under FIFRA. Devices are not subject to the registration requirements that apply to pesticides under FIFRA section 3. Pesticide devices must, however, be produced in a registered pesticide-producing establishment and that number must appear on the labeling of all pesticide devices.

EPA has identified many types of devices that are subject to FIFRA jurisdiction. Some aquatic-related devices include (but are not limited to) certain ultraviolet light systems, ozone generators, water filters and ultrasonic devices for which claims are made to kill, inactivate, entrap, or suppress the growth of pests in various sites. Aerators, nano-bubblers, water circulators and similar products which are marketed with claims to control algae, cyanobacteria or aquatic life in general, would all be considered to be pesticide devices. It has also been noted recently that devices utilizing fire, steam and lasers along with skimmers, vacuums, and the like – all claiming to manage or control aquatic pests – are making a comeback. They are still regulated as pesticide devices under FIFRA.

Harvesters and cookie cutters and similar devices are not regulated by the EPA even though they clearly are devices intended to manage aquatic plants. These types of devices fall under an exemption from regulation because the effectiveness of the device depends more on the performance of the operator than the performance of the devices itself. A flyswatter is another good example of a pesticide device exempt from regulation because its efficacy depends on the skill of the user and not the device itself.

The EPA also regulates the labeling of pesticide devices to some degree. In brief, the device is considered to be misbranded and subject to prosecution if the labeling fails to comply with the following requirements and others not listed here:

> The labeling bears any statements, designs, or graphic representations that are false or misleading;
> The label fails to bear the establishment number of the establishment where it was produced;
> It lacks adequate directions for use; or
> It lacks an adequate warning or caution statement.

While, as stated above, no registration of the device is required by the EPA, a manufacturer is barred from making any false or misleading claims for the device. In practice, that means, should the EPA ask for it, the manufacture must be able to satisfactorily prove with scientific evidence that their product does what it claims.

Use of unregistered pesticides for commercial purposes

As indicated above, products that directly or imply claims to reduce, control or manage plants, algae or cyanobacteria populations when used are thus considered to be pesticides. And “The claims ‘Reduces critical nutrients for cleaner, clearer ponds’, ‘Ponds with algae need to reduce nutrients’, and ‘Bacterial Product to Control Excess Nutrients for Clear, Clean Ponds’ imply pesticidal use and therefore require registration”. More and more often these days, companies or individuals will use a product known to reduce nutrients and either directly or by implications as described above, make claims to customers that the treatment will control a pest such as algae or cyanobacteria. Frequently such claims are associated with alum treatments, although there are other products used as well. The original manufacturer usually is not making claims. Rather, the user is the entity implying or actually claiming pesticide activity.

FIFRA Compliance Policy No. 3.5 states, in part:

> The Agency considers any application of an unregistered pesticide for other than personal use to be distribution or sale of an unregistered pesticide, a violation under Section 12(a)(1)(A) of FIFRA. This includes applying an unregistered pesticide to another person’s property for other than monetary consideration. Furthermore, a person applying an unregistered pesticide for hire, only to provide a service of controlling pests without delivering any unapplied pesticide to any person so served, would be considered a distributor and is therefore subject to the higher penalties set forth in section 14(a)(1) and 14(b)(1) of FIFRA.

The use of alum, lanthanum and other nutrient reducers, flocculants, etc., absent any additional claim, is perfectly legal and constitutes a viable option for water management. Once an expressed or implied claim to reduce, control or manage an aquatic organism has been made in association with the application, the applicator/operator is in clear violation of FIFRA. In addition, because the application of the unregistered pesticide is to, over, or near either waters of the U.S. or waters of the State, the provisions of the Clean Water Act are also triggered and compliance with that statute is also required.

Summary

The US Environmental Protection Agency was formed in 1970 and became responsible for regulating the rapidly expanding development and use of pesticides. During the course of the next 20 years, the use of some pesticides was cancelled, and testing requirements were developed to study the effects of pesticides on human health and the environment. These requirements are regularly revised to include the most recent developments in science. EPA toxicologists, chemists and biologists review proposed pesticide labels and revise label instructions as needed to ensure that human health and environmental safety will not be compromised. States may also register or approve pesticide labels for use in their jurisdictions and are allowed to add additional restrictions or requirements to the pesticide label. State guidelines cannot be less restrictive than those outlined on the federally approved label. The EPA and state regulatory agencies enforce pesticide laws regarding the purchase, use and disposal of pesticides. Pesticide labels are developed after years of research and include specific information about the pesticide and its use. The label is a legal document and all directions must be followed by those who use the product.

Photo and illustration credits:

Page 156: Herbicide testing; William Haller, University of Florida
Page 160: Aerator; William Haller, University of Florida
Page 161: Harvester; William Haller, University of Florida

3.7.1 Chemical Control of Aquatic Weeds

Jeffrey Schardt: Florida Fish and Wildlife Conservation Commission (retired), Thomasville GA; jeff.schardt@gmail.com
Michael D. Netherland: US Army Engineer Research and Development Center (deceased)

Introduction

Chemical control – the use of registered aquatic herbicides and algaecides – is a technique that is widely employed by aquatic plant managers in both private and public water bodies throughout the United States. Treatments can range in size from backpack spray applications for individual or small clusters of plants up to large-scale treatments from boats or helicopters that may target an invasive weed throughout an entire lake. While the objective of some treatments is broad spectrum control of numerous plant species, most herbicide applications target a specific invasive plant or algal species. The difference in scale, scope, timing, regulations and management objectives associated with the use of aquatic herbicides makes it a challenge to write an all-encompassing single document. In this section we seek to explain some of the rules and regulations associated with aquatic herbicide labeling; explain the difference between trade, chemical and common names; describe key differences between submersed and emergent applications; contrast contact and systemic herbicides; and provide specific information about each registered aquatic herbicide.

All herbicides discussed in this chapter have undergone EPA review (Section 3.7) and have been approved for aquatic use. This does not mean these herbicides are registered or can be used in every state since most states have their own regulatory and registration procedures. In addition, some states require applicators of aquatic herbicides to be certified or licensed before these products can be purchased and used. Many states also require that permits be obtained before herbicides can be applied to bodies of water – even if the waters are privately owned. Herbicide labels and SDS (safety data sheets) are available online on the registrant's website and are excellent sources of information. ***Always read the label on the herbicide and check with the appropriate regulatory agencies in your state before purchasing or applying pesticides to any body of water***.

Like all pesticides, aquatic herbicides have three names; a trade name, a common name and a chemical name. The trade name of a product is trademarked and is owned by the company, whereas the common name and the chemical name are assigned by the American National Standards Institute and the rules of organic chemistry, respectively. For example, consider the aquatic herbicide Rodeo®. The trade name of this herbicide is Rodeo®, the common name is glyphosate and the chemical name is N-(phosphono-methyl) glycine, isopropylamine salt. If a particular pesticide is protected by a patent, there may be only a single trade name associated with that pesticide. However, if the pesticide is off-patent, there may be multiple trade names that share the same common and chemical name. A number of aquatic herbicides are off-patent and have multiple trade names; therefore, we refer to herbicides by their common names only throughout most of this handbook.

There are approximately 300 herbicides registered in the US, but only 16 are currently registered for use in aquatic systems. Herbicide labels often include a list of the nuisance species controlled by the product, but applicators may be allowed to use the herbicide to control a target weed not listed on the herbicide label provided the product is labeled for use at the desired site of application. For example, if you wish to use an herbicide to control a weed in your pond and the weed is not listed on the herbicide label, you may still be able to use the product to control this particular weed if the label specifies that the herbicide may be used in ponds. However, it is important to check with state authorities before doing so because some states specify that herbicides can only be used to control weeds that are listed on the product label. Additionally, the user accepts liability for the performance of the product if the specific weed is not included on the label.

Herbicides can be classified in several ways, including by their chemical family, mechanism of action (specific site within the plant affected by the herbicide), mode of action (how they work and symptoms) and time of application in relation to growth of the weed. In this section we classify aquatic herbicides based upon how they are applied (as foliar or submersed treatments – although some herbicides are both) and on their activity in the plant (systemic or contact).

Products that are applied as foliar treatments are most easily recognized by the public. For example, if you have a weed to control, you select a herbicide based on label directions, mix the product with the prescribed amount of water and apply it directly to the weed. Contact products work quickly and kill the plant rapidly on contact (hence the designation "contact"). Systemic compounds, on the other hand, usually work slowly by affecting biochemical pathways and must be absorbed by the plant before providing control; therefore, systemic compounds may require days, weeks, or even months to kill the weed. The application method is the same for both systemic and contact herbicides – the compound is applied directly to the foliage of the plant. Foliar herbicides are used to control floating, floating-leaved and emergent aquatic weeds.

Submersed herbicides are applied as liquids, granules or pellets. Liquid treatments are often mixed with water to facilitate application and to ensure even distribution and are applied to achieve an entire water volume concentration to control submersed weeds or planktonic algae. Some dry formulations (wettable powders, water dispersible granules) are mixed with water and applied similar to liquids, but many granular and pelleted products are applied using granular spreaders. Aquatic herbicide applicators must determine the volume of the water to be treated before applying submersed herbicides to ensure that the appropriate and effective amount of herbicide is used. The following constants are needed to calculate the volume of water before treatment with submersed herbicides:

- The volume of a body of water is calculated in acre-feet, which is a function of area and depth; for example, a lake with an area of 1 acre and a depth of 6 feet has a volume of 6 acre-feet
- A single acre-foot of water comprises around 326,000 gallons of water and weighs around 2.7 million pounds

The volume (in acre-feet) of a water body, or treatment site within a water body, is used to determine the amount of herbicide needed to control a targeted weed for a submersed (or in-water) application. For example, if the label of a herbicide specifies an application rate of 1 ppm (part per million), then 2.7 pounds of the herbicide's active ingredient must be applied for each acre-foot of the water to effectively control the target weed. This results in a concentration of 1 ppm since 2.7 pounds of herbicide are mixed with 2.7 million pounds of water in each acre-foot. Most herbicide labels include a table that lists application rates, but it may be necessary to perform calculations similar to those described above to ensure that the correct herbicide dosage is applied. The labels of aquatic herbicides clearly state how these calculations are performed.

Mechanism of action vs. mode of action

The terms "mechanism of action" and "mode of action" are often used interchangeably. The mechanism of action is the actual biochemical site within the plant affected by the herbicide, whereas the mode of action describes the symptoms that occur after herbicide application leading to plant death. The difference is especially important in selecting herbicides for resistance management (Section 3.7.2). Knowing the mode of action is important to see and verify that plants are responding to an herbicide application. Understanding the mechanism of action allows managers to select an herbicide or combinations of herbicides that will provide acceptable plant management results while also providing some measure of resistance management. The Weed Science Society of America (WSSA) groups herbicides according to their mechanism of action providing managers with a better understanding of the potential for resistance development. The following tables list the mechanism and mode of action along with the WSSA Group for each aquatic-registered herbicide.

Contact herbicides

Several herbicides registered for aquatic use are classified as contact herbicides. This term may lead one to believe that these herbicides kill weeds immediately after contacting them. While contact herbicides tend to result in rapid injury and death of the contacted plant tissues, it is important to realize that the term "contact herbicide" refers to the lack of translocation or mobility of the herbicide in the plant after the herbicide is taken into the plant tissue. Herbicides that move through plant tissues following uptake are said to translocate; these products are called "systemic herbicides". This distinction between contact and systemic herbicides has significant implications for the prescribed use of the products and usually describes how quickly weeds may be controlled.

Contact herbicides are often used for foliar treatments of sensitive free-floating plants such as waterlettuce (Section 2.12), duckweed (Section 2.14) and salvinia (Section 2.13) and good spray coverage is essential to ensure control of all individual plants of these species. Contact herbicides are also used to temporarily control a number of emergent aquatic plants. These treatments are often initially effective but treating emergent plants with a contact herbicide often results in rapid recovery and significant regrowth from plant tissues that do not come into contact with the herbicide. As a result, systemic products are usually preferred for controlling emergent plants because systemic herbicides move or translocate within the plant and kill underground roots and rhizomes, which reduces or eliminates regrowth.

Contact herbicides that are used to control submersed weeds must remain in the water within the treated area for a few hours to a few days so that plants are exposed to a lethal concentration of the herbicide for a sufficient amount of time. The results of a herbicide application designed to control submersed plants is primarily impacted by two key factors:

1) the concentration of the herbicide in water that surrounds the target plant
2) the length of time a target plant is exposed to that herbicide before it dissipates or degrades

This dose/response phenomenon is herbicide- and plant-specific and has been defined as a concentration and exposure time (CET) relationship. Contact herbicides have relatively short exposure time requirements (often measured in hours or days), which means that these products are used to target specific areas within a larger water body or in areas where significant dilution is expected. Whether for contact or systemic herbicides, the vast majority of poor treatment results following submersed applications are due to an inability to maintain the herbicide in contact with the target plants at a lethal concentration for an appropriate period of time. Each contact herbicide has a different use rate, exposure requirement and selectivity spectrum. While the registered contact herbicides are often referred to as “broad-spectrum” products, there is a range of plant susceptibilities to each of these contact herbicides based on the species, use rate, treatment timing and exposure period. Proper identification of target and nontarget plants is important when selecting a contact herbicide because they can significantly differ in their selectivity to various plant species.

Susceptible submersed plants that are treated with contact herbicides typically show symptoms of herbicide damage within a day or two of treatment and collapse of the target plants can occur within 3 to 14 days. It is important to note that the use of contact herbicides in areas with dense plant populations and warm water temperatures can lead to a situation where decomposing plant tissue quickly depletes the oxygen from the water column, resulting in conditions that can cause a fish kill. Warm water holds much less dissolved oxygen than cool water. As microbes that decompose plant tissues increase, they can further consume already low oxygen levels below amounts needed for fish to breathe and survive. Product labels have directions that provide guidance to avoid oxygen depletion when treatments are made under conditions of dense vegetative cover and warmer water temperatures.

It is important that contact herbicides be applied and distributed as evenly as possible to the target plant (or throughout the water column for control of submersed plants) to ensure that the entire plant – including the rooted portions of the plant near the sediment – is exposed to the herbicide. Poor mixing of contact herbicides within the water column can result in control of plant tissue growing near the water surface, followed by rapid recovery from the lower portions of the plant that were not exposed to the herbicide. Poor control can also result from summer applications when treating lakes that are thermally stratified (Section 3.7.4).

Contact herbicides are currently used for both small-scale treatments such as along shorelines and for large-scale control efforts. Most of the contact herbicides have been registered for many decades and tend to be versatile with a wide range of use patterns. Combinations of two or more contact herbicides are often used to target specific invasive or nuisance species. The registered contact herbicides (and dates of registration) are described in more detail below. These brief descriptions are not comprehensive but are meant to serve as a guide to historical strengths or potential issues associated with the use of these products.

Compound Date registered for aquatic use	Primary use: Submersed	Primary use: Floating	Primary use: Emergent	Formulation	Mechanism of action (MOA) WSSA resistance management group	Mode of action / comments
Copper 1950s	X	X		Liquid chelates Granular $CuSO_4$ Granular chelates	Plant cell toxicant WSSA Group is undefined	- Contact herbicide / algaecide - Used alone or in combination with other herbicides - Often used for submersed plant control near potable water intakes - Typical use rate 0.2 to 1 ppm
Endothall 1960	X	X		Liquid Granular	Inhibits protein phosphatase (PP1) enzyme WSSA Group is undefined	- Fast-acting contact herbicide primarily for submersed weeds - Treatment timing affects selectivity - Dipotassium salt for submersed plant control (typical use rate 2 to 3 ppm) - Increased use in irrigation canals (2010) - Dimethyl-alkylamine salt for algae and plants that are more herbicide-tolerant (typical use rate 0.3 to 3 ppm)
Diquat 1962	X	X	X	Liquid	Photosystem 1 inhibitor WSSA Group 22	- Contact, broad spectrum herbicide - Inhibits photosynthesis and destroys cell membranes - Turbidity affects effectiveness - Very fast activity on sensitive plants; faster activity under high light conditions - Typical use rate 0.1 to 0.37 ppm
Peroxides 2002		X		Liquid Granular	Oxidizes algal cell membranes WSSA Group is undefined	- Contact algaecide - Affects cell wall permeability, cell membrane integrity - Algae control, particularly cyanobacteria and certain species of filamentous algae
Carfentrazone 2004	X	X	X	Liquid	Inhibits a plant-specific enzyme (PPO) WSSA Group 14	- Contact herbicide - Causes rapid desiccation and necrosis - Waterlettuce and broadleaf weed control - Activity on select submersed species - pH of the water can impact efficacy - Typical submersed rate 50 to 200 ppm - Typical foliar rate 4 oz per ac
Flumioxazin 2011	X	X	X	Liquid Water dispersible granule	Inhibits a plant-specific enzyme (PPO) WSSA Group 14	- Contact herbicide - Causes rapid desiccation and necrosis - Waterlettuce, surface sprays for algae control, submersed plant control - pH of the water can impact efficacy - Typical submersed rate 50 to 200 ppb - Typical foliar rate 4 to 8 oz per ac - Mixed with glyphosate to control some emergent plants

Copper (1950s)

Copper is a micronutrient that is needed for healthy growth of animals and is often added to animal feed and to vitamins formulated for human use. Copper is widely used as a fungicide in agricultural systems to control diseases on food crops and copper-based compounds have been used for aquatic plant control since the early 1900s. Copper is a fast-acting, broad-spectrum, contact herbicide that kills a wide range of algae (Section 2.18) and aquatic plants primarily used in potable waters, irrigation canals, ponds, lakes and reservoirs. Copper sulfate is likely the most widely used copper product. Submersed use rates typically range from 0.2 to 1.0 ppm copper in the water column. There are no restrictions on the use of copper in potable water sources or in waters used for crop irrigation. This allows the immediate use of treated water.

Copper acts quickly on plants and algae and has a short exposure requirement, which can be advantageous when treating small areas or areas subject to rapid dilution. High alkalinity or hard water can reduce the effectiveness of copper-based products. Chelated copper compounds were developed in the 1970s to address these problems. Chelate is a chemistry term meaning combining a metal ion, in this case, copper, with an organic molecule, triethanolamine or ethylenediamine. Chelated liquid copper products reportedly remain in solution longer than copper salts when applied to hard water. Copper that is in solution (suspended in the water) for a longer time has greater effect on the aquatic plants and algae. Copper chelates are broken down by hydrolysis and rapidly decline to ambient concentrations with a half-life in water of 2 to 8 days depending on conditions.

All copper formulations are considered toxic to mollusks and can be toxic to some species of fish at relatively low doses, especially if the water has less than 50 ppm of carbonate hardness (soft water). Levels of 1 to 5 ppm are toxic to fish, so copper is applied for aquatic weed and algae control at concentrations of 1.0 ppm or less. Toxicity generally decreases as water hardness increases. Copper does not biodegrade, and regular use can result in increased copper residues in the sediment. Copper is generally considered to be biologically inactive once bound in the sediments.

Endothall (1960)

Endothall acid, first available as an aquatic herbicide in the 1960s, was originally used in agriculture as a plant desiccant. In aquatic sites, endothall is used primarily to control submersed plants and use rates and methods of application vary widely. Endothall is fast-acting and is treated as a contact herbicide but it may be somewhat mobile in plant tissues. Endothall is absorbed by submersed plants in lethal concentrations in 12 to 36 hours depending on the concentration applied. Endothall acid works by interfering with plant respiration, affecting protein and lipid biosynthesis and disrupting plant cell membranes. It causes cellular breakdown of plants within 2 to 5 days. Symptoms of plant damage, including defoliation and brown shriveled tissues, will become apparent within a week of herbicide application. Plants will fall out of the water column within 3 to 4 weeks after application.

Traditional use patterns of endothall have included spot treatments of small target areas generally applied at the highest label rate and where species selectivity is not a major concern. Selective use is based on species sensitivity, use rates and treatment timing. The effectiveness of endothall is generally not affected by factors such as alkalinity or turbidity of the water. In recent years, large-scale early-season treatments of the dipotassium salt formulation of endothall have been applied to target invasive plants such as hydrilla (Section 2.2), curlyleaf pondweed (Section 2.4) and Eurasian watermilfoil (Section 2.3) that persist throughout the winter. These treatments are conducted before desirable native plants begin to grow in spring, which may allow control of the invasive weeds with limited impact on native species that grow later in the season. It is important to note that these early-season treatments are applied when plant biomass is not at its peak and when water temperatures are cooler. These conditions reduce or prevent oxygen depletion that may occur when fast-acting herbicides are applied to dense nuisance populations of weeds in warmer water. Early season, large-scale application rates are generally effective at concentrations as low as 2 to 3 ppm. It is important to treat the entire target area as quickly as possible to avoid herbicide loss through dilution or degradation. Endothall is also widely used in control of submersed weeds and algae in irrigation canals and has irrigation restrictions.

Diquat (1962)

Diquat is a fast-acting contact herbicide that is rapidly absorbed by plant tissues and interferes with photosynthesis in susceptible plant species. Diquat effectively controls many free-floating weeds via foliar applications including duckweed, watermeal (Section 2.14), waterlettuce, waterhyacinth (Section 2.11) and salvinia. Diquat kills the aerial portions of plants in 24 to 36 hours, which occurs too quickly to allow translocation to other parts of the plant. As noted above, good coverage is critical because missing even a small area or a few individuals can lead to rapid recolonization by these fast-growing floating species. Diquat is also used to control submersed plants in small treatment areas or in areas where dilution may reduce the time that plants are exposed to the herbicide. It diffuses rapidly through the water column during submersed applications and is quickly absorbed into plant tissue.

Diquat is generally considered to be a “broad-spectrum” product that kills a wide range of plant species. However, the susceptibility of different submersed species can vary significantly. Diquat can be rapidly inactivated when treating “muddy” or turbid water and the speed of this inactivation can interfere with plant control. There are no hard and fast rules to determine when water is too muddy to treat, but the effectiveness of diquat increases as water clarity increases. Diquat is often mixed with copper-based herbicides to control a broader range of weeds and to improve control of target plants. It can also be applied with low rates of endothall to reduce exposure time needed to control some submersed plants, notably hydrilla. In addition to its use in aquatic systems, diquat is labeled for weed control in turf and along fence lines and has been used to desiccate the leaves and vines of potato to increase ease of harvesting.

Peroxides (2002)

Sodium carbonate peroxyhydrate is a granular or liquid substance registered for use in aquatic systems for control of planktonic algae (Section 2.18), especially blue-green algae (also known as cyanobacteria). This contact algaecide is used mainly for control of problematic algae including in domestic water supply sources, with very limited use for control of submersed vascular plants. Sodium carbonate peroxyhydrate rapidly transforms via hydrolysis into hydrogen peroxide and sodium carbonate. Hydrogen peroxide is the active component. It works by oxidizing critical algal cellular components resulting in rapid membrane disruption and death. Hydrogen peroxide then rapidly breaks down into water, oxygen and other natural products while the sodium carbonate component is degraded to sodium and bicarbonate ions. The half-life for this process is approximately 8 hours. Blowers or granular spreaders are used to ensure uniform coverage of the water surface when using granular formulations. Best results typically occur when treatments are applied on sunny days prior to the onset of a significant algal bloom. Often used to control algae in potable water supplies, hydrogen peroxide is also widely used in the medical field to kill bacteria. The use of peroxides for submersed plant control has been investigated and is generally not considered to be effective.

Carfentrazone (2004)

Like a systemic herbicide, carfentrazone affects a plant-specific enzyme, in this case protoporphyrinogen oxidase (PPO); however, the rapid onset of symptoms (membrane destruction, tissue necrosis) is similar to contact herbicides. In contrast to the contact herbicides mentioned above, carfentrazone has a much narrower spectrum of weed control. While this can limit the utility of the product to a few target weeds, it can also result in improved selectivity and reduced damage to nontarget plants. Carfentrazone is absorbed through the leaves and inhibits the PPO enzyme that is important in chlorophyll synthesis. It needs 1 to 2 hours of contact for good herbicidal activity, but is fairly slow-acting once inside the leaves, causing symptoms in 2 to 5 days and plant necrosis (tissue death) in about 3 to 4 weeks.

Carfentrazone breaks down both through microbial action in soil and through hydrolysis with a half-life of 3 to 5 days in water. It has very low toxicity to fish and waterfowl. Carfentrazone has been used for control of waterlettuce and duckweed, and in combination with other herbicides for selective control of some broadleaf emergent plants. It provides good selectivity since it will not control non-target plants like pickerelweed (*Pontederia cordata*) or grasses that may be mixed with targeted invasive species like waterlettuce. Combinations of carfentrazone and imazamox have provided good control of large-flowered primrosewillow (*Ludwigia grandiflora*). Carfentrazone is also labeled for submersed plant control. However, limited use to date has hampered the development of new use patterns for this product and more research is needed before it will be widely used on submersed weeds. Managers have noted that carfentrazone performance improves when applications are made on sunny days, but high pH waters may reduce carfentrazone activity on submersed plants due to rapid degradation of the herbicide. Carfentrazone is also used for weed control in turf, corn and other crops.

Flumioxazin (2011)

Flumioxazin was registered by the EPA for aquatic use in 2011. It is a contact herbicide with the same mechanism of action as carfentrazone and the onset of rapid injury is similar to other contact herbicides. However, flumioxazin has a broader spectrum of activity compared to carfentrazone. Like carfentrazone, flumioxazin is a PPO enzyme inhibitor. It moves within treated leaves but does not translocate to other areas of the plant. Flumioxazin disrupts plant cell membranes and the result is cell leakage, inhibited photosynthesis, bleaching of the chloroplasts and cell death. Plant necrosis and death is rapid, taking a few days to a week or two. In general, at least 4 hours of contact time is required for good control. The primary breakdown pathway of flumioxazin in water is by hydrolysis and is highly dependent on water pH. Under high pH values (greater than 9), flumioxazin half-life in water is 15 to 20 minutes. Under more neutral pH values (7 to 8), the half-life in water is around 24 hours.

Flumioxazin controls a wide variety of aquatic weeds and algal species. Submersed plants include Eurasian watermilfoil, cabomba (Section 2.6) and hydrilla. Floating waterlettuce, duckweeds, giant salvinia, watermeal and surface mats of some algae are susceptible to flumioxazin. Typical application rates range from 50 to 200 ppb for submersed plants and 4 to 8 oz per acre applied as a foliar spray for floating plants. Flumioxazin use patterns are still being developed, but rates as low as 2 to 4 oz per acre (foliar spray) or 50 ppb (submersed application) are reportedly effective for waterlettuce control. Field evaluations have shown that surface and submersed flumioxazin applications also provide control of the native species spatterdock (*Nuphar* sp.), waterlily (*Nymphaea* sp.) and American lotus (*Nelumbo lutea*). Flumioxazin can be tank-mixed with other contact or systemic herbicides to enhance control of emergent weeds like large-flowered primrosewillow when used in combination with glyphosate, imazamox or auxin mimic herbicides.

Systemic herbicides – auxin mimics

In contrast to contact herbicides, systemic herbicides are mobile in plant tissue and move through the plant's water-conducting system (xylem) or food-transporting vessels (phloem). Once the herbicide is absorbed into the plant, it can move through one or both of these vessels and throughout the plant tissue to affect all portions of the plant, including underground roots and rhizomes. Auxin mimic herbicides simulate auxin, a naturally occurring plant hormone that regulates plant growth. These herbicides generally target broadleaf plants (dicotyledons or dicots) and are often called "selective herbicides" because many aquatic species (particularly grasses or monocots) are not susceptible to auxin mimic herbicides. Most submersed aquatic plants are monocots, which aids in selectivity when using an auxin mimic. After treatment, the shoot tissue of susceptible plants will often bend and twist (epinasty) and plants will frequently collapse within 2 to 3 weeks. Like contact herbicides, auxin mimics that are used to control submersed weeds must remain in the treated area for a few hours to a few days so that plants are exposed to a lethal concentration of the herbicide for a sufficient amount of time. Exposure times of 12 hours or greater may provide good control if the application rate and timing are appropriate. Longer exposure periods (such as 24 to 144 hours) increase the probability that the target weed will be completely controlled. The contact herbicides discussed above are used to control a large number of nuisance and invasive plant species. Auxin mimic herbicides are used for control of a much smaller number of plant species.

Compound Date registered for aquatic use	Primary use			Formulation	Mechanism of action (MOA) WSSA resistance management group	Mode of action / comments
	Submersed	Floating	Emergent			
2,4-D Granular ester - 1959 Liquid amine - 1976 Granular amine - 2009 Liquid acid – 2004	X	X	X	Granular ester and amine Liquid amine and acid	Auxin mimic, plant growth regulator WSSA Group 4	- Systemic herbicide - Absorbs into leaves; moves to growth areas - Selective broadleaf plant control - Used for submersed dicots such as Eurasian watermilfoil and for floating waterhyacinth management - Typical submersed rate 0.5 to 4 ppm - Typical foliar rate 2 to 4 pounds per ac
Triclopyr 2002	X	X	X	Liquid amine Liquid acid	Auxin mimic, plant growth regulator WSSA Group 4	- Systemic herbicide - Absorbs into leaves; moves to growth areas - Used for submersed dicots such as Eurasian watermilfoil and for floating and emergent plants - Typical submersed rate 0.25 to 2.5 ppm - Typical foliar rate 1 to 3 pounds per ac
Florpyrauxifen-benzyl 2018	X	X	X	Liquid: Soluble concentrate (SC) Emulsifiable concentrate (EC)	Auxin mimic, plant growth regulator WSSA Group 4	- Systemic herbicide - New mechanism of action for aquatic plant control - Targets a specific receptor in plants - Selective floating, emergent and submersed plant control - Typical submersed rate 2 to 48 ppb - Typical foliar rate 2.7 fl oz/ac (0.026 to 0.052 pounds a.i. per ac)

While there are several aquatic dicotyledons (and some monocots) that show sensitivity to the auxin mimics, these herbicides have historically been used for selective control of a limited number of emergent, floating and submersed plants, including purple loosestrife (Section 2.16), waterhyacinth and Eurasian watermilfoil. The auxin mimics 2,4–D and triclopyr have very similar use patterns and are used to control broadleaf plants growing among desirable grasses or native submersed plants. This is referred to as "selective control" and is very important in natural aquatic sites to maintain native species while reducing growth of invasive weeds. These herbicides are also widely used to control weeds in turf, pastures, forestry and other terrestrial sites.

2,4–D (1959)

2,4-D (2,4-dichlorophenoxyacetic acid) is the oldest organic herbicide registered in the US and was developed to increase crop yields during World War II. It is also the first selective herbicide since it controls broadleaf plants but usually does not control grasses. It is primarily used for weed control in food crops (grains, corn, sorghum, rice, sugarcane), turf, non-crop areas and in certain aquatic environments. 2,4-D was first applied in aquatic systems to control waterhyacinth in the 1950s. It is a systemic, auxin mimic herbicide that is absorbed by roots and leaves, then translocates and accumulates mainly in the growing points of shoots and roots. 2,4-D interferes with the plant's ability to maintain proper hormone balance. Plants undergo uncontrolled growth in some tissues and halted growth in other tissues. The result is injury to the growing regions of the plant and then a gradual death, usually within 1 to 3 weeks.

Several nuisance emergent and submersed plants are controlled by 2,4–D, but this herbicide is primarily used for selective control of waterhyacinth and Eurasian watermilfoil. A liquid amine formulation is used to control emergent and submersed plants and a granular ester formulation is used for submersed weed control. In addition, a granular amine and a low volatile acid formulation have been recently registered. 2,4-D is applied alone or mixed with other herbicides like diquat, flumioxazin or glyphosate to improve efficacy or resistance management. Some native emergent plants – including waterlilies, spatterdock and bulrush – are susceptible to 2,4–D, so care should be taken to avoid injury to these plants. Microbial degradation is the primary breakdown pathway for 2,4-D, resulting in a half-life that ranges from one to several weeks. The half-life is shorter in warmer months and in waters to which 2,4-D has been previously applied, presumably where microbial activity is greater.

2,4-D is sometimes confused with "Agent Orange", a name given to the military's plant defoliant mixture that was developed and used during the Vietnam War. During the manufacture of Agent Orange (2,4-D mixed with 2,4,5-T), it was contaminated with cancer-causing dioxin (tetrachlorodibenzo-p-dioxin), known as TCDD. Although 2,4-D was one of the components of Agent Orange, it is not Agent Orange, nor does it contain TCDD, nor has it been shown to cause cancer. After numerous lifetime feeding studies in rats and mice, the EPA has categorized 2,4-D as Class D compound (Not Classifiable as to Human Carcinogenicity).

Triclopyr (2002)

Triclopyr has been widely used to control herbaceous and woody plants in non-cropland sites, forestry and pastures. It was registered for aquatic use in 2002. Similar to 2,4–D, several plant species are susceptible to triclopyr; however, the historical strength of this auxin mimic herbicide has been selective control of invasives such as Eurasian watermilfoil or purple loosestrife. Triclopyr is absorbed by foliage and translocates throughout the plant. It moves to areas of new growth and causes a disruption in hormone levels, interfering with normal expansion and division of plant cells. It acts like a growth stimulant in some plant tissues and a growth retardant in others. Symptoms include cupped leaves and twisted stems. Vascular tissue becomes crushed, stopping movement of essential nutrients and sugars, and plants essentially grow themselves to death.

Photolysis is the primary breakdown pathway of triclopyr in water. Triclopyr has a short half-life depending on season and water depth (e.g. 2.5 days in warm shallow water during the summer to 14 days in cooler deeper water in winter). Triclopyr does not bind strongly or adsorb to soil particles. It is registered as both liquid and granular amine salt and liquid acid formulations. Some native non-target emergent plants are more susceptible to triclopyr than they are to 2,4-D, so care should be taken to avoid injury to these plants.

Florpyrauxifen-benzyl (2018)

Florpyrauxifen-benzyl is used for broadleaf and grass control in flooded rice fields and was registered by the EPA for aquatic use in 2018. It has been assigned reduced risk status, is practically non-toxic to fish and wildlife and has no drinking, fishing or swimming restrictions. Florpyrauxifen-benzyl represents a new mechanism of action and offers unique properties for aquatic plant control as a member of the arylpicolinate herbicide chemical family. Florpyrauxifen-benzyl is a systemic herbicide with an affinity for aquatic vegetation. It is an auxin mimic that is absorbed through leaves and shoots via foliar treatments or underwater tissues during in-water applications. Following rapid uptake, the herbicide translocates in the xylem and phloem and accumulates in meristematic tissues where it bonds with a specific target receptor. It mimics the effect of a persistent high dose of the natural plant hormone auxin, causing overstimulation of specific auxin-regulated genes, which results in the disruption of several growth processes in susceptible plants. Susceptible plants become brittle and shatter within a few days after exposure. Plants exhibit reduced growth and ultimately turn chlorotic and necrotic, reaching a level of control within 1 to 3 weeks after treatment.

The primary breakdown pathway for florpyrauxifen-benzyl in water is by photolysis. The half-life in shallow water is less than 12 hours and 1 to 2 days in deeper water. Breakdown is slightly enhanced via hydrolysis in waters with pH values greater than 8. Very high turbidity or algal content may subtly reduce florpyrauxifen-benzyl uptake by target aquatic weeds following in-water application due to the herbicide's strong binding to organic matter and merit the use of higher rates within the label range. Florpyrauxifen-benzyl is a highly active herbicide and controls susceptible plants at very low application rates or concentrations. For example, foliar rates for susceptible emergent and floating plants are only 0.026 to 0.052 pounds of active ingredient per acre; in contrast, other Group 4 herbicides are often applied at 1 or more pounds per acre. The auxin herbicides have long been used to control Eurasian watermilfoil and this species is controlled by florpyrauxifen-benzyl at concentrations of 8 ppb or lower. Hydrilla, which is not usually affected by auxin herbicides, is susceptible to florpyrauxifen-benzyl and is controlled at concentrations of 20 to 48 ppb. Floating, floating-leaved and emergent weeds such as floatingheart (Section 2.9), waterprimrose (*Ludwigia* spp.) and waterhyacinth are also listed as susceptible on the product label. Because florpyrauxifen-benzyl is active at such low concentrations, care must be taken to avoid damage to susceptible native or non-target plants and irrigation restrictions must be followed. This product was recently registered for aquatic use so additional data and use patterns are being evaluated in several current and planned field operations.

Systemic herbicides – enzyme specific herbicides for foliar use

The herbicides glyphosate and imazapyr are not labeled for submersed use (both break down rapidly in water and thus are not effective for submersed weed control); instead, they are labeled only for foliar treatment and control of emergent and floating plants. They are systemic and readily move through plant tissue to control aboveground and underground portions of emergent plants. These herbicides target enzymes in pathways found only in plants and they inhibit enzymes that are required for growth, so treated plants slowly "starve" and eventually die. Herbicides that target plant-specific enzymes typically show very low toxicity to non-plant organisms such as mammals, fish and invertebrates. Glyphosate and imazapyr herbicides are truly broad-spectrum, and a very limited number of emergent plant species can tolerate exposure to them. Glyphosate and imazapyr are especially effective at controlling large monotypic stands of invasive and nuisance emergent plants such as phragmites (Section 2.15), cattail (*Typha* sp.) and other perennial plants that have extensive rhizome and root systems. Both herbicides result in fairly slow control of target weeds and are often mixed together for plants that are particularly hard to control.

Compound Date registered for aquatic use	Primary use			Formulation	Mechanism of action (MOA) WSSA resistance management group	Mode of action / comments
	Submersed	Floating	Emergent			
Glyphosate 1977			X	Liquid	Inhibits plant-specific enzyme (EPSPS) WSSA Group 9	- Systemic herbicide - Broad spectrum for emergent plant control - New growth stunted - Plant death may be slow - Not active in soil or in water
Imazapyr 2003			X	Liquid	Inhibits plant-specific enzyme (ALS) WSSA Group 2	- Systemic herbicide - Broad-spectrum for emergent plant control - Plant death may be slow - Active in soil – cannot be used in irrigation ditches

Glyphosate (1977)

Glyphosate is a broad-spectrum, systemic herbicide used to control annual and perennial broadleaf weeds, grasses, trees and certain floating plants. It is widely used in agriculture, homeowner and specialty markets, including aquatics. Glyphosate is translocated through treated plant tissues. It works by interfering with the plant-specific shikimic acid pathway, which produces the enzyme EPSPS, and inhibiting the synthesis of specialized plant amino acids. Without the ability to manufacture these essential components, plant death occurs slowly over a period of 2 to 3 weeks. Animals do not produce these enzymes, which accounts for the very low toxicity of this herbicide to animals. Visible effects on most annual weeds occur within 4 to 7 days (longer on most perennial weeds), and 30 days or more on most woody plants and trees. Glyphosate has no soil activity and is rapidly deactivated in natural waters via binding to various cations in the water. Therefore, it cannot be used for control of submersed weeds. Because this herbicide is rendered inactive so quickly, the irrigation and potable water restrictions associated with the use of glyphosate are minimal.

Treatment timing can impact the effectiveness of glyphosate and nuisance species should be treated during late summer or fall when plants are moving sugars to storage organs such as roots or rhizomes in preparation for overwintering. This treatment timing can increase the translocation of glyphosate into the storage organs and often results in enhanced control of the target plant during the following growing season. Glyphosate breaks down in water microbially with a variable half-life of 12 to 60 days. It binds readily with soil or suspended organic particles, effectively inactivating its herbicidal properties. Also, hard water (containing calcium or magnesium cations) used to make a spray solution may bind some of the glyphosate in the mix tank prior to application. Selective management of plants using glyphosate is achieved only by careful application because, in general, glyphosate damages most plants it contacts. There are an increasing number of glyphosate-resistant weeds reported in agricultural settings. Therefore, it is important to rotate or combine herbicides with different MOAs to the extent possible when applying glyphosate.

Imazapyr (2003)

Imazapyr was discovered in the 1970s. It is used in forestry and specialty markets, including aquatics, where it was registered for control of aquatic weeds in 2003. Imazapyr is a systemic herbicide that is quickly absorbed by leaves and shoots and moves to areas of new growth where it shuts down plant growth almost immediately. In this regard, imazapyr acts like a contact herbicide. It inhibits the production of the plant-specific acetolactate synthase (ALS) enzyme; plants are not able to grow without this enzyme, so they eventually starve and die. Susceptible plants become reddish at the growing tips within 1 to 2 weeks and control may take 2 to 6 weeks. Imazapyr is absorbed through plant leaves and roots and should be applied when target plants are actively growing in the spring, summer or fall. If applied to mature plants, a higher concentration of herbicide and a longer contact time will be required.

Imazapyr has been used to control invasive plants such as spartina, phragmites and other perennial weeds that have invaded previously unvegetated areas in tidal zones or river flats. Imazapyr is also applied alone or with glyphosate to control cattail and tussocks (floating masses of herbaceous and woody species). Unlike glyphosate, imazapyr is active in the soil so care should be taken to avoid treating areas around the root zones of desirable plants, particularly near trees along the water's edge. Imazapyr is practically non-toxic (the EPA's lowest toxicity category) to fish, invertebrates, birds and mammals. The primary degradation pathway for imazapyr in water is via sunlight with a half-life of about a week or less. Breakdown is via microbial degradation in terrestrial soils and may take weeks to months.

Systemic bleaching herbicides

Compound Date registered for aquatic use	Primary use			Formulation	Mechanism of action (MOA) WSSA resistance management group	Mode of action / comments
	Submersed	Floating	Emergent			
Fluridone 1986	X	X		Liquid Granular	Inhibits plant-specific enzyme (PDS) WSSA Group 12	- Systemic herbicide - New shoot growth is bleached - Large-scale or whole-lake management - Low use rates, long exposure requirements - Treatment timing and rate affect selectivity - Used for some floating plants - Typical submersed rate 5 to 30 ppb
Topramezone 2013	X	X		Liquid	Inhibits plant-specific enzyme (HPPD) WSSA Group 27	- Systemic herbicide - New shoot growth is bleached - Large-scale or whole-lake management - Low use rates, long exposure requirements - Treatment timing and rate affects selectivity - Used to control some floating plants - Typical submersed rate 20 to 40 ppb

Fluridone (1986)

Fluridone is a systemic herbicide that was discovered in the mid-1970s and was initially used for weed control in cotton. It was later shown to be effective for the control of submersed aquatic plants and was registered by the EPA for aquatic use in 1986. Fluridone is a bleaching herbicide that targets the plant-specific enzyme phytoene desaturase (PDS), which protects chlorophyll (the green pigment responsible for photosynthesis in plants) from damage by UV light. Fluridone is used primarily to control submersed [e.g., Eurasian watermilfoil, hydrilla and egeria (Section 2.5)] and floating (e.g., duckweed, watermeal and salvinia) plants by treating the water column. Fluridone symptoms are highly visible, with the new growth of sensitive plants bleaching and turning pink or white as chlorophyll in the plant is destroyed by sunlight. Susceptible plants show bleaching symptoms in new shoot growth; however, it is important to note that bleaching symptoms don't always equal control and actual plant death may not occur for weeks or months after an initial treatment.

Fluridone can be both selective and broad-spectrum and use rates vary from 4 to 150 ppb, but rates of 12 to 40 ppb are most commonly used. Higher rates often provide broad-spectrum control, whereas lower rates increase selectivity. Unlike the contact or auxin mimic herbicides that require hours or days of exposure, the fluridone label states that target weeds must be exposed to fluridone for a minimum of 45 days. The extended exposure requirement typically calls for treatment of the entire aquatic system or treatment of protected embayments of lakes or reservoirs. Required exposure periods depend on the plant species, stage of plant growth and treatment timing. During the exposure period, new shoot growth of susceptible plants becomes bleached and this continuous bleaching of new growth depletes the plant's reserves of carbohydrates needed for growth. This slow death (which may take two or more months) can allow plants to continue to provide structure for habitat and produce oxygen through photosynthesis. There are limited restrictions for potable water use and no restrictions on fishing or swimming; however, irrigation restrictions are described on the product label. Fluridone is available in several liquid and pellet formulations. All formulations require that plants be exposed to sufficient concentrations of fluridone for an appropriate period of time. As a result, sequential fluridone treatments – often called "bumps" – are usually applied several weeks or more than a month apart to ensure that an effective concentration of the herbicide is maintained. This is especially important for controlling submersed plants. Due to the long-lived nature and critical exposure time requirements, periodic sampling is often conducted to measure fluridone concentrations in treated water and to determine whether further applications are necessary to maintain a lethal concentration of the herbicide in the water. Floating plants are generally controlled much more quickly than submersed species. Fluridone can be used in systems ranging from less than one acre to several thousand acres.

The main degradation pathway for fluridone is via sunlight, but fluridone is also broken down by microbes. Resistance to this herbicide was confirmed at several research institutions in 2000 after repeated failures of large-scale fluridone applications for hydrilla control in Florida in the late 1990s. This was the first occurrence of resistance to a bleaching-type herbicide and the first for a plant species based solely on somatic mutations (Section 3.7.2). As mentioned in Section 2.2, hydrilla reproduces solely by vegetative means (asexually, no seed production) in Florida, leaving no avenue for gene recombination between different plants. Fluridone attacks only one gene location in hydrilla and several genotypes (plants with a specific unique mutation in that gene) have since been reported in Florida, with each genotype having a different level of resistance to fluridone. Repeated fluridone use effectively removed the susceptible hydrilla genotypes, leaving plants with the resistance mutations to expand and become dominant.

Topramezone (2013)

Topramezone has been used to control broadleaf and grass species in corn as well as in conifer and other non-crop areas. It was registered for aquatic use by the EPA in 2013. Topramezone is the first aquatic-registered herbicide belonging to the chemical class called pyrazolones and can be used in rotation with other modes of action in resistance management programs. It is a systemic herbicide that is applied to the water column for submersed or floating plant control, or directly to foliage of floating and emergent vegetation. In sensitive plant species, topramezone inhibits the enzyme 4-hydroxy-phenyl-pyruvate-dioxygenase (4-HPPD), which leads to a disruption of the synthesis and function of chloroplasts. Consequently, chlorophyll is destroyed by oxidation, resulting in bleaching symptoms (white or pink coloration) of the growing shoot tissue within 7 to 10 days after exposure and subsequent death of the plant.

Topramezone shares many of the characteristics described for fluridone. These include: 1) low use rates (20 to 40 ppb); 2) extended exposure requirement of greater than 45 days; 3) rate-based selectivity; 4) bleaching of new plant growth; 5) slow death of target plants; 6) water sampling to manage long-term herbicide concentrations; 7) no use restrictions on drinking, swimming and fishing; and 8) whole-lake or large-scale use patterns. The current topramezone label includes submersed weeds such as hydrilla and Eurasian watermilfoil and some floating plants. Applications are made to actively growing plants early in the growing season before mature plants can build carbohydrate reserves, mat at the water surface, and slow growth and subsequent herbicide response. Applying early in the growth stage reduces the amount of herbicide and the exposure period necessary to control hydrilla. Waterhyacinth has been controlled via root uptake of topramezone in waters treated for hydrilla control. The main degradation pathway for topramezone is via photolysis with a half-life in water ranging from 4 to 6 weeks. Microbial degradation is a minor breakdown pathway for topramezone, which may also weakly adhere to suspended clay particles.

Systemic herbicides – ALS herbicides

Several recently registered herbicides include compounds that target the plant-specific enzyme acetolactate synthase (ALS). As noted above for imazapyr, ALS inhibitors stop the production of three amino acids (isoleucine, leucine and valine) which then inhibits the production of ALS enzymes and other proteins that are built from these amino acids. Although the exact mechanism is not understood, when ALS is inhibited, plants die. Animals do not produce these enzymes, so penoxsulam has low toxicity to animals. In contrast to the broad-spectrum activity described for glyphosate and imazapyr, the ALS herbicides tend to be much more selective. Despite a similar mode of action, use patterns vary substantially among the ALS products. Similar to other enzyme specific inhibitors, these herbicides are applied at comparatively low use rates and result in a slow kill of the target weed. Susceptible floating plants are often controlled much more quickly than large emergent rooted plants or submersed plants. Although systemic ALS herbicides do not result in bleaching of new plant growth, they are similar to the bleaching herbicides. They require 1 to 3 or more months of exposure to achieve control of submersed weeds since the plants slowly deplete their carbohydrate reserves.

Compound Date registered for aquatic use	Primary use			Formulation	Mechanism of action (MOA) WSSA resistance management group	Mode of action / comments
	Submersed	Floating	Emergent			
Penoxsulam 2007	X	X		Liquid	Inhibits plant-specific enzyme (ALS) WSSA Group 2	- Systemic herbicide - New growth stunted - Large-scale control of hydrilla and other submersed plants - Extended exposure required for submersed plant control - Typical submersed rate 10 to 30 ppb - Floating plant control - Typical foliar rate 2 to 5.6 oz/ac
Imazamox 2008	X	X	X	Liquid Granular	Inhibits plant-specific enzyme (ALS) WSSA Group 2	- Systemic herbicide - New growth stunted - Selective emergent plant control - Typical foliar rate 32 to 128 oz/ac - Growth regulation and control in hydrilla - Typical submersed rate 25 to 75 ppb
Bispyribac-sodium 2012	X	X		Wettable powder	Inhibits plant-specific enzyme (ALS) WSSA Group 2	- Systemic herbicide - New growth stunted - Large-scale control of hydrilla and other submersed plants - Extended exposure required for submersed plant control - Typical submersed rate 20 to 40 ppb - Floating plant control - Typical foliar rate 1 to 2 oz/ac

Penoxsulam (2007)

Penoxsulam was originally registered in 2004 for broadleaf, grass and sedge control in rice and turf. It was registered for aquatic use in 2007 and is currently used to manage floating species including waterhyacinth, waterlettuce and salvinia and submersed plants such as hydrilla. Treatments may include foliar application of penoxsulam directly to the target floating plants or submersed application for control of both submersed and floating plants. Penoxsulam is a systemic herbicide that is absorbed by plant tissues and moves to areas of new growth. During the exposure period, new shoot growth is inhibited and plants can turn red in color due to stress. The extended exposure requirement typically necessitates treatment of the entire aquatic system or application to protected embayments of lakes or reservoirs where dilution from water exchange is minimized. Despite the extended herbicide contact time associated with penoxsulam treatments, there are no restrictions on use of water for drinking, fishing or swimming, but irrigation restrictions are described on the product label.

Penoxsulam is broken down primarily via photolysis and to a lesser extent by microbes. Its half-life in waters ranges from 7 to more than 30 days and is dependent on water depth and light intensity. Enzyme inhibiting herbicides act very slowly and control is highly dependent on contact time. Penoxsulam use rates and exposure requirements for submersed applications are generally similar to those of fluridone, topramezone and bispyribac-sodium and plant death may occur over a period of 60 to 100 or more days depending on the plant species, stage of plant growth and treatment timing.

This may necessitate split or multiple applications to keep the herbicide concentration at at 20 to 40 ppb for up to 100 days for optimum performance.

Resistance to ALS herbicides has been reported in terrestrial plants, so mixing these products with a second active ingredient with a different mechanism of action can reduce the likelihood of resistance development. Combining penoxsulam with the dipotassium salt formulation of endothall substantially decreases the exposure time needed to control hydrilla to about 7 to 14 days, which reduces the effects of degradation and dissipation and the need for additional applications to maintain appropriate penoxsulam concentrations in the water column. Applying these herbicides in combination provides a measure of resistance management, requires less of each herbicide to control hydrilla and may increase selectivity to conserve non-target native plants. Likewise, applying penoxsulam in combination with flumioxazin or carfentrazone has provided an effective waterhyacinth management tool with increased resistance management benefit over penoxsulam alone.

Imazamox (2008)

Imazamox was registered by the EPA in 1997 for broadleaf and grass control and has been used in agricultural sites including soybeans, rice, sunflowers and wheat. It was registered for aquatic use in 2008. Imazamox is a systemic herbicide that is applied to plant foliage to control floating or emergent plants, or to the water for submersed plant control. Imazamox is broken down in the water by photolysis and microbial degradation and its half-life in water is 7 to 14 days.

Imazamox is available in liquid and granular formulations and should be applied to actively growing plants, preferably early in the season to young plants with lower carbohydrate reserves. It is rapidly absorbed into plant tissues and growth of susceptible plants is generally inhibited within a few hours after application. In this regard, it acts somewhat like a contact-type herbicide, requiring a short exposure period. However, plant death is relatively slow. Meristematic tissues become chlorotic in 1 to 2 weeks; this is followed by general chlorosis and death in 2 to 6 weeks. Primary aquatic plant management uses of imazamox include foliar applications to control cattail, wild taro (*Colocasia esculenta*), Uruguayan primrose willow complex (*Ludwigia grandiflora/hexapetala*) and waterhyacinth. Imazamox may be applied alone or in combination with other herbicides like glyphosate or carfentrazone. Applying imazamox alone provides a measure of selectivity for comingled non-target plants. Combining with glyphosate or carfentrazone results in more rapid and thorough control but is usually limited to monocultural stands of target plants or other areas where selectivity is not a concern. Imazamox may provide up to a year of control of waterhyacinth via root uptake from in-water applications.

Bispyribac-sodium (2012)

Bispyribac-sodium has been labeled and used for weed control in rice for many years and was registered by the EPA for aquatic plant control in 2012. It is a systemic herbicide that is absorbed into and moves within the affected plant. Like most systemic aquatic herbicides, control is highly dependent on contact time or exposure of the plant to the herbicide. The primary degradation pathway for bispyribac-sodium is microbial metabolism and the half-life in water is about 30 days.

Bispyribac-sodium is applied alone via subsurface injection at 30 to 45 ppb to manage hydrilla and requires 60 to 90 days of exposure to achieve control. With a half-life in water of about 30 days, monitoring and reapplication to maintain the original desired concentration and exposure period without exceeding the label maximum of 45 ppb. Combining lower rates of bispyribac-sodium with as little as 1 ppm of dipotassium salt of endothall not only reduces the exposure time required to control hydrilla, but also provides a measure of resistance management by utilizing two different mechanisms of action.

These three ALS-inhibiting herbicides clearly demonstrate that each herbicide is different since the use rates and the spectrum of plants controlled are very different, particularly between imazamox and the other two herbicides in this section.

Herbicide dissipation, degradation, deactivation and half-life

The length of time a herbicide remains in contact with target plants following a submersed application is critical to achieving desired results. Two key processes that dictate the required exposure of plants to herbicides are dispersion and degradation. Once applied to the water, herbicides are subject to dispersion or movement both within and away from the treated area. Dispersion initially has a positive influence on the treatment because it facilitates mixing of the herbicide in the water column. The rate of movement of herbicide residues from the treatment area is likely the largest single factor affecting treatment success, especially for treatments applied to a small area in a large water body. For example, application of a herbicide to a 10-acre protected cove in a large reservoir may result in limited movement outside the treatment area and a subsequent long exposure period. In contrast, a 10-acre plot applied along an unprotected shoreline of the same reservoir on the same day may result in the herbicide moving out of the target area and becoming diluted to less-than-lethal concentrations within a few hours of treatment. Conditions on the day of treatment can be very important, especially for treatments applied to unprotected areas of larger lakes. High winds or high water flow associated with recent precipitation can have a strong negative influence on treatment results. Since the potential range of exposure periods can vary significantly at the same site from day to day, even greater variation between sites is likely. This variation in the expected exposure period will often influence both choice and application rate of the selected herbicide.

In addition to dispersion, herbicide degradation plays a significant role in the effectiveness of a treatment. Except for copper (a natural element), all herbicides are subject to degradation pathways that ultimately lead to breakdown products that include carbon, hydrogen and other simple compounds. These degradation pathways result in decomposition of the herbicide to simpler products that lack herbicidal activity via processes such as photolysis (breakdown by ultraviolet rays in sunlight), microbial degradation (breakdown via action of the microbial community) or hydrolysis (breakdown via the action of water splitting the herbicide molecule). Environmental conditions such as temperature, hours of sunlight, trophic status of the water body (Section 1.1) and pH can all influence the rate of degradation of the different herbicides. In terms of herbicidal effectiveness, degradation pathways are particularly important for products like fluridone, penoxsulam, topramezone or bispyribac-sodium that require long exposure periods of 45 to 100 or more days. In these situations, the entire water body is often treated and therefore dispersion or dilution is not an issue, but the rate of degradation will often dictate product effectiveness and may require additional “bump” treatments to maintain lethal concentrations for the required exposure times.

The role of pH for products that are degraded via hydrolysis such as flumioxazin and carfentrazone is a relatively new factor in aquatic plant management and managers need to consider pH as a significant factor in product performance. It is also important to mention the phenomenon of herbicide deactivation in relation to herbicide effectiveness. Several herbicides can bind with various ions in the water column, which can result in a reduction or loss of herbicidal activity. Binding is not a degradation pathway, but it can have an important influence on herbicide effectiveness. The best examples of product binding are the immediate binding of glyphosate to positively charged cations in the water column and the binding of diquat to negatively charged particles such as clay or organic matter in the water column. In both cases, the herbicide molecule remains intact but no longer has any herbicidal activity. The bound particles eventually settle to the sediments where microbial degradation takes place. Herbicides that are chemically bound in the sediment no longer have herbicidal activity and undergo microbial degradation over time.

The tables on pages 177 and 178 provide general information about exposure time requirements, typical aqueous half-lives that result from product degradation and the key degradation pathways for aquatic herbicides.

Herbicide concentration monitoring

The above discussion of herbicide dissipation and half-lives is relevant to current use patterns of many aquatic herbicides. Operational monitoring of herbicide concentrations has increased significantly over the past 10 years. The advent of enzyme-linked immunoassays (ELISA) for several of the registered aquatic herbicides (including fluridone, endothall, triclopyr, 2,4–D, penoxsulam and bispyribac-sodium) has largely been responsible for this trend. While monitoring used to be very costly and was associated almost exclusively with regulatory studies or field research trials, several groups now offer monitoring support for operational treatments. When managers select herbicides such as fluridone and bispyribac-sodium, the extended exposure requirements and large-scale use patterns are often supported by monitoring programs. In this case, monitoring can be used to manage the concentrations and exposure periods and to determine when and if additional herbicide applications are necessary to achieve optimal target plant control. In addition, monitoring can be used to determine when herbicide concentrations become low enough that use restrictions on water can be lifted (e.g. irrigation, potable water use). There are numerous potential uses for operational monitoring of aquatic herbicide concentrations; given the lower analytical costs and the value of the information that can be obtained, it is likely this trend will increase in the future.

Summary

This chapter lists 16 chemicals that are registered by the EPA for aquatic plant control in aquatic systems. These herbicides are very different from one another; some have been used for decades, whereas others have only recently been approved for use in water. More specific directions regarding the use of these products are on the label and are also available from the companies that manufacture, sell or distribute these herbicides.

Contact herbicides: contact exposure requirements, half-lives and degradation pathways

Compound	General exposure requirements	Typical half-life in water	Key degradation pathway and comments
Copper	Hours to 1 day	Hours to 1+ day	Copper is a natural element and is therefore not subject to degradation. Following application, copper ions are typically bound to particles or chemical ions in the water or sediment which results in the loss of biological activity. Active copper ions in the water column are more readily inactivated in hard water systems. Concerns have been expressed regarding buildup of copper residues in sediments.
Endothall	Hours to 2-3 days	2 to 14+ days	Endothall is a simple acid that is degraded via microbial action. Water temperature and the level of microbial activity can have a strong influence on the rate of degradation. Cooler water temperatures typically result in slower rates of degradation.
Diquat	Hours to days	0.5 to 7 days	Diquat is rapidly bound to negatively charged particles in the water column. Higher turbidity water can result in very fast deactivation of the diquat molecule. The ionic bonds between diquat and charged particles negate herbicidal activity. Once biologically inactivated, diquat is then slowly degraded via microbial action.
Peroxides	Minutes to hours	Rapid ~8 hours	Peroxide based algaecides are short-lived in the water column and quickly breakdown via biotic and abiotic processes including hydrolysis. Degradation is enhanced in warm alkaline waters.
Carfentrazone	Hours to 1+ day	Hours to 3 to 5 days	Carfentrazone is degraded via hydrolysis. The rate of hydrolysis is pH–dependent, with faster degradation occurring in higher pH waters.
Flumioxazin	Hours to 1+ day	Minutes to 1+ day	Flumioxazin is degraded via hydrolysis and the half-life has been calculated as ~5 days, 24 hours, and 22 minutes at pH of 5, 7, and 9 respectively. The pH has a strong influence on efficacy of flumioxazin.

Systemic herbicides: contact exposure requirements, half-lives and degradation pathways

Compound	General exposure requirements	Typical half-life in water	Key degradation pathway and comments
2,4-D	Hours to days	4 to 21+ days	The key degradation pathway for 2,4-D is via microbial action. Warmer water temperatures increase the rate of microbial activity and can have a strong influence on the rate of degradation. Photolysis also plays a role in degradation.
Triclopyr	Hours to days	3 to 14+ days	The key degradation pathway for triclopyr is via photolysis or sunlight. Time of year, water depth and water clarity influence the rate of photodegradation. There is also some microbial action that results in degradation.
Florpyrauxifen-benzyl	Hours	0.5 to 2 days	The key degradation pathway for florpyrauxifen-benzyl in water is by photolysis or sunlight. Breakdown is faster in shallower or clearer water. Breakdown is slightly enhanced via hydrolysis in basic (pH > 8) waters.
Glyphosate	Several hours to 1 day	Hours to 1+ day to inactivate Degradation in soil: 12 to 60 days	Glyphosate is rapidly deactivated once it contacts the water column due to immediate binding with positively charged ions in the water. Once bound to cations, glyphosate is biologically inactive. Microbial action ultimately degrades the glyphosate molecule in the sediment.
Imazapyr	At least 1 hour for foliar applications Not used for submersed	7 to 14+ days	The key aqueous degradation pathway for imazapyr is via photolysis. Time of year, water depth and water clarity can influence the rate of photodegradation. Microbial degradation can also play a role.
Fluridone	45+ days	7 to 30+ days	The key degradation pathway for fluridone is via photolysis. Factors such as water depth, water clarity and season of application can influence photolytic degradation. Microbial activity can also play an important role in degradation.
Topramezone	45+ days	14 to 30+ days	The key degradation pathway for topramezone is via photolysis. Factors such as water depth, water clarity and season of application can influence the rate of photolytic degradation. Microbial activity can also play a supporting role in degradation.
Penoxsulam	45+ days	7 to 30+ days	The key degradation pathway for penoxsulam is via photolysis. Factors such as water depth, water clarity and season of application can influence photolytic degradation. Microbial activity can also play a supporting role in degradation.
Imazamox	Several hours	7 to 14+ days	The key degradation pathway for imazamox is via photolysis. Factors such as water depth, water clarity and season of application can influence photolytic degradation. Microbial activity can also play a supporting role in degradation.
Bispyribac-sodium	45+ days	30+ days	Bispyribac-sodium is degraded via microbial action. Factors such as water temperature, trophic status, and plant density can influence the rate of degradation. Bispyribac-sodium generally has a long half-life; however, faster rates of degradation have been noted in a limited number of sites.

3.7.2 Herbicide Resistance & Resistance Management of Aquatic Plants

Ryan A. Thum and Gregory M. Chorak: Montana State University, Bozeman MT; ryan.thum@montana.edu, gregory.chorak@student.montana.edu

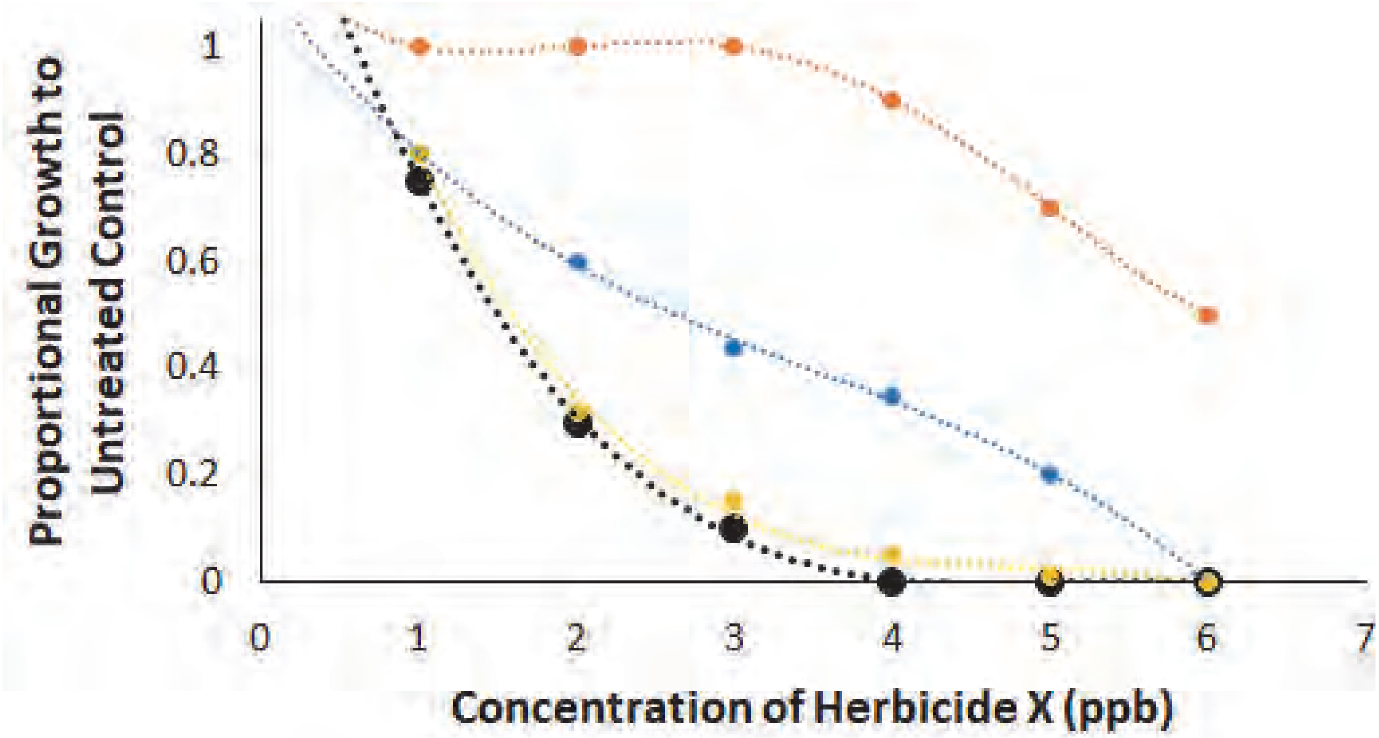
What is resistance?

The accepted definition of herbicide resistance is "the inherited ability of a plant to survive and reproduce following exposure to a dose of herbicide normally lethal to the wild type. In a plant, resistance may be naturally occurring or induced by such techniques as genetic engineering or selection of variants produced by tissue culture or mutagenesis". In the following paragraphs, we dissect this definition into its key terms.

The term "inherited" means the trait is passed down from parents to offspring, at least in part. This means that offspring tend to resemble their parents because they inherit their genes from their parents. More specifically, this means that variation among individuals for some phenotypic trait (e.g., how a plant responds to different doses of an herbicide) arises partly because those individuals possess different genes that influence the trait. This is in contrast to variation among individuals that arises solely from environmental factors, such as how much light or nutrients they receive, or whether or not they are under attack by natural enemies such as herbivores or pathogens.

Geneticists measure inheritance (also referred to as heritability) in different ways (see Thum 2018 listed in the additional information section of this handbook for more information). Since many aquatic plants can reproduce clonally, heritability can be demonstrated and estimated as differences among clones (genotypes) in a trait of interest (i.e., phenotype) under common conditions. For example, hybrid watermilfoil genotypes have heritable differences in their response to 2,4-D. Similarly, a hybrid watermilfoil genotype from Townline Lake in Michigan has been vegetatively propagated and has repeatedly shown a reduced response to fluridone compared to other genotypes, so we can confidently conclude that the Townline Lake fluridone response is inherited. Heritability can also be demonstrated if specific DNA sequences can be shown to determine particular traits of interest, since organisms pass their DNA to their offspring. An example of this is the mutations in the phytoene desaturase gene that confer different levels of fluridone resistance in hydrilla (Section 2.2). However, since the genes that determine most phenotypes are unknown, this method is uncommon and it is important to recognize that inheritance can be demonstrated without knowing the specific gene(s) involved in determining a phenotype.

Next, let's break down the phrase "dose of herbicide normally lethal to the wild type". A "wild type" is the typical prevailing characteristics of a species under natural conditions. However, atypical or mutant types can be found in most species. For example, most of us think of the familiar gray squirrel as having a grayish-brown coat on top with a white coat on bottom. However, all-black or all-white squirrels of this same species are sometimes seen. In this case, the grayish-brown squirrels are the wild type and the black or white squirrels are mutants with respect to the wild type. For a given trait, as long as there is a reference for what constitutes a normal, expected response of a species to an herbicide, then any herbicide response that is elevated over that reference would be considered resistance. For example, assume that 4 parts per billion (ppb) of herbicide X is normally lethal to the wild type for a species; a specific genotype of that species that can survive and reproduce when exposed to 4 ppb of herbicide X would be considered resistant, so long as it can be demonstrated that the ability to survive and reproduce at four ppb of herbicide X can be passed down from parents to offspring.



Example of genetic variation for herbicide response. The wild type (large black circles) is normally killed by four parts per billion of herbicide X. However, other genotypes have different responses. In some cases, response to the herbicide may only be slightly elevated for a different genotype (small yellow circles). However, other genotypes may exhibit a high level of resistance at concentrations normally lethal to the wild type (small blue circles and small orange circles).

Resistance arises because mutations occur naturally and randomly when DNA replicates. In some cases, these mutations happen to provide an advantage in survival and/or growth of a plant in the presence of a certain amount of an herbicide. Thus, when a population is exposed to the herbicide, the individuals that have mutations conferring some level of resistance are more likely to survive and reproduce than the wild type individuals. Over time, the population can become dominated by the mutant genotype(s) and these genotypes could also spread to other lakes.

Herbicide tolerance

Herbicide tolerance is defined as "the inherent ability of a species to survive and reproduce after herbicide treatment. This implies that there was no selection or genetic manipulation to make the plant tolerant; it is naturally tolerant". A key difference between resistance and tolerance is that the latter is a characteristic "of a species". For example, many monocots are naturally tolerant to doses of 2,4-D that impact Eurasian watermilfoil (Section 2.3) and therefore repeated use of 2,4-D may result in increased abundances of these naturally tolerant species. Therefore, the repeated use of a given herbicide may result in a shift in species composition of an aquatic plant community towards species that are naturally tolerant to that herbicide. This phenomenon is not the same as resistance, because resistance arises from genetic variation among individuals within a species that has a susceptible wild type. Nevertheless, the impacts of selecting for tolerant species via herbicide use can be of management concern.

Examples of herbicide resistance in aquatic plants

As of this writing, there are currently 512 cases of documented weed resistance to 167 different herbicides (International Survey of Herbicide Resistant Weeds, http://www.weedscience.org; accessed 28 May 2020). Most documented cases of herbicide resistant weeds come from terrestrial agriculture but several aquatic plants are resistant to herbicides used in rice agriculture in Australia and Asia. For example, populations of starfruit (*Damasonium minus*), dirty dora (*Cyperus difformis*) and arrowhead (*Sagittaria montevidensis*) have evolved resistance to bensulfuron. Pervasive weed resistance in agricultural systems understandably fuels concerns regarding herbicide resistance in aquatic plant management of private and public waters.

Herbicide resistance in aquatic plants is rare compared to the large number of cases documented in terrestrial agriculture. The most well-known case of herbicide resistance in aquatic plants is fluridone resistance in dioecious hydrilla, but a population of duckweed (*Landoltia punctata*; see Section 3.14) in a Florida canal was highly resistant to diquat and paraquat compared to wild type duckweed. Also, as mentioned earlier, hybrid watermilfoil from Townline Lake in Michigan has developed resistance to fluridone that is not seen in other Eurasian and hybrid watermilfoil genotypes.

The dearth of documented cases of herbicide resistance in aquatic plants in the US begs the question: is herbicide resistance in aquatic plants truly rare or is it present but not tested for or reported enough? It is possible that factors promoting herbicide resistance (see below) are commonly lacking in aquatic plant management, leading to very low occurrences of resistance evolution. However, it is also possible that reduced efficacy in some populations goes undetected because of a lack of quantitative pre- and post-treatment monitoring or because explanations for reduced efficacy do not consider the possibility of resistance (e.g., are explained by environmental factors that may have limited the dosage of herbicide). Thus, it is important to quantitatively monitor herbicide efficacy and to consider herbicide resistance when efficacy is lower than expected. Conclusive testing for resistance would then come from laboratory studies comparing suspected resistant types to known wild type (susceptible) genotypes.

Factors that may influence the development of herbicide resistance

It is important to recognize that there are different physiological and genetic mechanisms for herbicide resistance. **Target-site resistance** refers to mutations that lead to changes in the molecule(s) where the herbicide typically binds, which leading to less effective binding and thus less impact on the weed. For example, hydrilla that is resistant to fluridone has amino acid changes in the phytoene desaturase gene. It is generally thought that herbicides with a single site of action will be more prone to the evolution of resistance, since mutations occurring at the target enzyme can directly confer resistance. It is therefore important to note that many of the herbicides registered for aquatic plant control target a single enzyme. In contrast, **non-target site resistance** refers to genetic changes that occur not at the herbicide's site of action, but at other genes that are related to herbicide uptake, translocation, sequestering, detoxification or metabolism. Whether herbicide resistance results from target versus non-target site mutations should be contingent upon the supply of mutations available within a population when a herbicide is applied.

Genetic mechanisms for herbicide resistance can also be broadly classified into **single-gene** versus **polygenic** (multiple genes). In single-gene resistance, the level of resistance observed is conferred by mutation at a single gene, which would most likely occur at the target site. However, resistance can occur via changes at multiple genes, each of which confers some fraction of the overall resistance observed. Thus, a given level of resistance could reflect mutations at a single gene that have a large effect on resistance or by mutations at many genes that each have a small effect on resistance.

In general, high rates of herbicides should select for mutations in a single gene that have a large effect and low rates of herbicides should select for polygenic resistance [because at low rates and in genetically diverse populations there may be alleles (different forms of a trait) at different genes that can each allow higher survivorship and reproduction at low rates]. For example, some individuals may have genes that allow them to metabolize small amounts of a particular herbicide, so they can continue to grow in the presence of low doses of herbicide, while other individuals may have genes that help them to sequester small amounts of that herbicide. The population of individuals that survive treatment with that particular herbicide are then enriched for these two different genes, both of which confer a small degree of herbicide resistance (e.g., survival and continued growth at low doses). Subsequent intercrossing of individuals with these two different genes can allow "gene stacking" that confers resistance to a higher dose of herbicide to offspring than either parent can tolerate because the new genotypes can both metabolize and sequester the herbicide.

Examples of this phenomenon can be found in the terrestrial agricultural literature but it is unknown whether or how commonly this occurs in aquatic plants. Certainly, the exposure of plants to low rates of herbicides is inevitable in many aquatic plant management projects that utilize spot treatments in large water bodies where the herbicide will rapidly dissipate. Gene stacking of low-level resistance alleles will depend on the extent of genetic variation within and among populations and the extent to which sexual reproduction occurs. Thus, different control tactics (e.g., whole lake applications where long contact times are maintained vs. spot applications that likely dissipate to sublethal doses) may interact with plant life histories (e.g., asexual, sexual or mixed reproductive strategies) to influence the probability and magnitude of herbicide resistance. Strong academic research on this issue could be helpful in the future for building better models of when, where and how resistance could occur, as well as developing efficient tools to detect it.

Resistance management

Traditionally, the idea behind resistance management is to prevent or delay the evolution of resistance. Some commonly used methods of resistance management are listed below.

Rotations and/or combinations of herbicides with different modes of action can limit resistance evolution. Individuals are less likely to be resistant to all of the herbicides in the mixture or rotation because resistance to different herbicides will likely require independent mutations in different target genes. However, rotations and combinations do not guarantee that resistance will not evolve. If populations are large enough, it is possible (although unlikely) that an individual will have multiple mutations conferring resistance to multiple herbicides. Similarly, individuals that harbor different mutations at different target genes can intercross and produce offspring with mutations at all of the different target genes. Further, non-target site resistance mutations that influence uptake, translocation, metabolism or detoxification may confer resistance to multiple herbicides (see http://www.weedscience.org).

Another resistance management strategy is to allow a sufficient amount of time in between applications of the same herbicide. This idea is based on the premise that resistant genotypes that survive treatment may exhibit a trade-off whereby they are less competitive in the absence of the herbicide (i.e., there is a “cost” to resistance), so allowing time to pass between treatments should cause the number of resistant individuals to go down if there is a cost to being resistant in the untreated environment. However, it is important to recognize that the success of this strategy depends on sufficient costs of resistance, which isn’t necessarily true; also, it isn’t clear how long in between treatments is sufficient. Therefore, this strategy relies on having detailed information on the level of resistance, costs, and dynamics of competition over time.

One method that is used to reduce resistance evolution in agricultural systems is to maintain untreated populations adjacent to treated populations, with the hope that interbreeding with an untreated population where there is no selection for resistance will keep resistance genes from becoming dominant in the treated population. This strategy has not been intentionally implemented in aquatic plant management to our knowledge. However, it is possible that this strategy is implemented de facto because across the landscape there is a mosaic of treated and untreated lakes. Furthermore, spot treatments are common within many lakes, which may ultimately have this same effect. However, it is generally thought that sexual reproduction plays a small role in the overall reproduction of most managed aquatic plants, so it is unclear whether the intentional implementation of this strategy would work, unless resistant genotypes are frequently replaced by wild type genotypes due to a cost of resistance as described above.

In addition to resistance management strategies, it is important that managers implement best management practices to maximize the impact of any control implementation. First, early detection and rapid response methods are recommended so that populations are treated when they are small and before they become a problem. The probability that a population harbors a resistance mutation will be proportional to its size, and therefore small populations are less likely to harbor a resistance mutation unless the population was initiated by a resistant genotype to begin with (e.g., colonized from a nearby source that is resistant). Second, it is generally recommended to treat with the maximum allowable herbicide rate to minimize the number of surviving plants. Finally, management should include quantitative monitoring efforts so that surviving plants are identified and targeted for appropriate follow-up management and so that changes in efficacy over time can be identified (see below).

Limitations to implementing resistance management in aquatic plant control operations

Most stakeholders in aquatic plant management recognize the potential for herbicide resistance and therefore the benefits of practices that prevent or delay the evolution of resistance. However, it is also important to recognize that there are logistical realities that limit the implementation of resistance management practices in many locations. The most challenging conditions under which to practice resistance management are likely to occur in large, public, multiple use water bodies where numerous factors influence the control options that are available. In these water bodies, the choice of herbicide, dose and timing is influenced by a variety of factors, including balancing different uses of the water body, cost of management, selectivity and efficacy and hydrology. Given the limited number of herbicides available for aquatic use, resistance management practices that involve rotations or mixtures of different products may be infeasible in certain water bodies and options may be limited to non-herbicidal techniques.

A second limitation to resistance management practices is justification for implementing them if there is no evidence that repeated use of a herbicide in a water body is selecting for resistant genotypes. It remains to be seen whether the risk of herbicide resistance in aquatic plant management is high, and to date there are few documented examples of resistance. The evolution of resistance will be limited by the supply of mutations available to confer enhanced survival and reproduction after exposure to herbicides. Thus, herbicide resistance involves a "waiting time" for mutations, which may or may not occur over the life of a management project. Given the regulatory demands for demonstrating selectivity and efficacy, managers may be resistant to adopting resistance management strategies if they have no evidence that their favored strategy and tactics for a water body are deemed efficacious (Section 3.1), especially if alternatives employed in resistance management practices are more costly and/or have less public support for their use.

Finally, a geographic region may represent a mosaic of water bodies that do and do not implement resistance management practices. In this case, it would be reasonable for managers that voluntarily implement resistance management practices (or are required to do so) at additional costs to ask themselves why they are doing it if others are not. Furthermore, because many aquatic plants can spread vegetatively among water bodies, locations with resistance management practices could potentially be colonized by resistant genotypes from other areas that do not practice resistance management. Therefore, it is important to be on the lookout for resistance and understand its origins even when resistance management is practiced.

Recommendations for addressing the potential for herbicide resistance

As a practical matter then, how should aquatic plant managers address the issue of resistance? Managers should consider all available options to implement resistance management practices wherever and whenever possible. However, the likely reality is that in many situations, resistance management practices will be limited. An immediate priority should be to develop and implement methods that objectively and definitively identify whether control efficacy is lower than expected and the likelihood that reduced efficacy is due to the presence of resistant genotypes.

Lake management plans (Section 3.2) should have monitoring protocols that can objectively and definitively separate out factors influencing the variation in control efficacy. For example, temporal data on control efficacy when the same control tactic is repeatedly employed should be informative for identifying reduced efficacy over time within a water body. In such a case, laboratory determinations of dose-responses to the current practice and alternatives could be used to determine whether a change in management strategy is warranted. This kind of objective data could provide compelling evidence to warrant changes in management strategies or tactics that may otherwise be resisted due to increased costs, regulatory hurdles or unfavorable public opinion.

Genetic tools hold promise to increase the efficiency in which we are able to test for resistant genotypes and identify resistance evolution. Since resistance evolution occurs as the displacement of genotypes that have wild type sensitivity to herbicides by mutant genotypes that are resistant, genetic information could assist in the identification of resistance. More specifically, we expect to see shifts in the genetic composition of populations over time if resistance is developing within a water body. Moreover, since most aquatic plants reproduce primarily by asexual means (e.g., fragments, turions, etc.), we may expect lakes that have quantitative data demonstrating lower than expected control efficacy to be dominated by a single clone. Thus, these genetic signatures can be used to prompt laboratory dose-response studies of specific genotypes suspected of exhibiting herbicide resistance.

Genetic tools currently available for hydrilla and Eurasian watermilfoil illustrate the promise of these tools. For example, fluridone-resistant biotypes of hydrilla can be identified with genetic data, which can help managers determine the dose of fluridone required for efficacy given the genetic composition of the population or whether fluridone is a poor control option for that particular population. Similarly, a fluridone-resistant genotype has been identified in hybrid Eurasian watermilfoil; molecular markers can distinguish this genotype from other genotypes and have shown that this genotype has spread to several populations in Michigan. Similar to hydrilla, genetic surveys of hybrid Eurasian watermilfoil can be used to determine whether fluridone should be applied to particular lakes. Characterization of genotypes in other species could be employed in the same way.

Summary

Wild type (normal) plants are susceptible to a particular herbicide, while other genotypes of the same species may become resistant to that herbicide as a result of mutations or intercrossing. There are a number of tactics to reduce the likelihood of developing herbicide resistance in populations of aquatic weeds but options available to managers can be limited based on the situation. Rotating or combining herbicides with different modes of action may limit resistance, and genetic testing may be very useful to guide management decisions regarding herbicide selection.

Photo and illustration credits:

Page 179: Hydrilla bouquet; William Haller, University of Florida
Page 180: Genetic variation for herbicide response; Ryan Thum, Montana State University
Page 181: Landoltia duckweed; Ben Willis, SePRO Corporation
Page 183: Hydrilla tubers (white) and turions (green); Lyn Gettys, University of Florida

3.7.3 Spray Adjuvants: A User's Guide

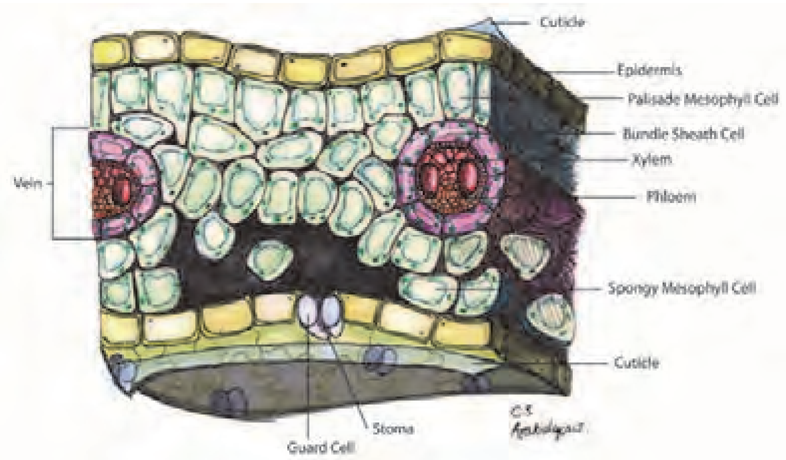
Jason Ferrell: University of Florida, Gainesville FL; jferrell@ufl.edu

Introduction

There are a number of herbicide brands on the market, but most products are applied either as specially formulated herbicide pellets (or granules) or as a liquid spray applied to water or plant stems and foliage. Of these techniques, spraying the foliage of undesirable plants is by far the most common practice. A spray adjuvant will usually be added for foliar applications to improve herbicide performance. However, there are hundreds of different adjuvant products available, so confusion often abounds and applicators may use products they don't need or fail to use products that could be helpful.

What is an adjuvant?

According to the Weed Science Society of America, an adjuvant is "any substance in an herbicide formulation or added to the spray tank to modify herbicidal activity or application characteristics" (Herbicide Handbook – 9th edition). There are two concepts that should be drawn from this definition: 1) an adjuvant is <u>not</u> herbicidal in and of itself, but rather works <u>with</u> the herbicide to improve efficacy, and 2) some adjuvants are used simply to improve the application and handling characteristics of a given herbicide. With this in mind, adjuvants are commonly divided into two primary categories: <u>activator</u> adjuvants and <u>utility</u> adjuvants. Activator adjuvants improve herbicide retention on and absorption into the leaf, while utility adjuvants are used to reduce spray drift, foaming in the tank and other factors not directly related to herbicide absorption or penetration into the plant.



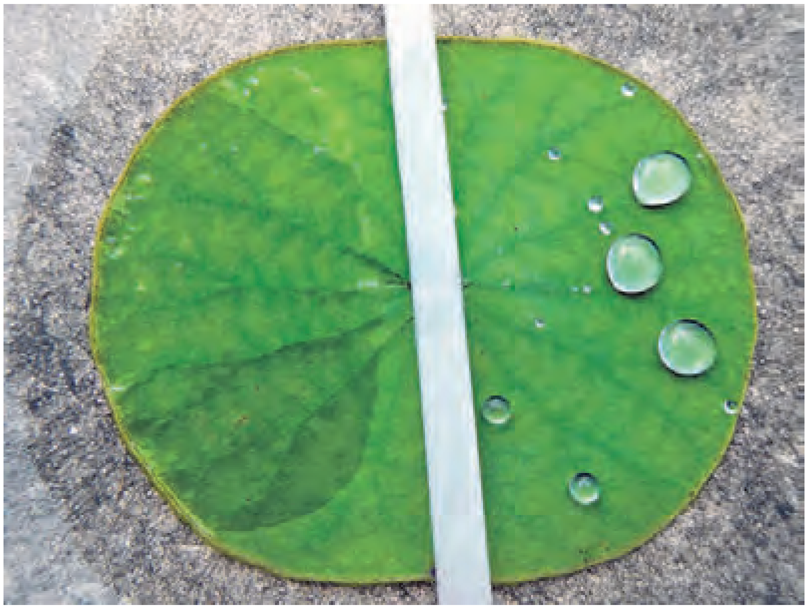
Before we talk about how different adjuvants work, we should first examine a plant leaf to understand how herbicides are absorbed into a typical emergent or terrestrial plant. This leaf cross section shows many different tissue and cell types, but of particular interest are the large veins in the middle of the leaf. These veins contain the xylem and phloem, which are specialized tissues that transport water and nutrients throughout the plant. Many herbicides (such as glyphosate) are highly effective because they are systemic, meaning they are moved in the phloem throughout the entire plant and result in total kill. But to kill the plant, these herbicides must first reach the veins in order to be transported. This is no easy task since the herbicide must land on the leaf, diffuse through the tissues, and reach the active site at a high enough concentration to be lethal. The active site is the location in the plant where herbicides interfere with enzyme production or other biochemical pathways to kill the plant. Both sides of the leaf are covered in a layer of wax called the cuticle. The cuticle is important to the leaf, since wax repels water and prevents it from "leaking" out of the leaf. Most foliar herbicides are diluted in water, so the cuticle is a formidable barrier to herbicide entry into the plant.

Activator adjuvants

As stated previously, activator adjuvants do not have herbicidal properties, but rather work with the herbicide to improve efficacy. The primary role of an activator adjuvant is to help the herbicide breach the cuticle barrier and enter the leaf. This group of adjuvants is often further divided into two broad categories: 1) wetter/spreaders, also generically called surfactants, and 2) penetrants.

Wetter/spreaders

Wetter/spreaders are often called surfactants or stickers and are likely the most common type of adjuvant used to improve herbicide performance. Members of this class, which are specially developed soaps, are quite effective while also being inexpensive. Their main function is to not interact with the herbicide per se, but to change the properties of the spray mixture in order to increase movement of the herbicide into the plant.



Why is this important? Recall that the leaf's waxy cuticle repels water. At the same time, molecules of water are attracted to each other, which causes them to form round, bead-shaped droplets (think raindrops). When no surfactant is added to a spray solution, the absorption of herbicide into the leaf is limited for two reasons. First, the round, bead-like droplet prefers to stay as a round droplet. Therefore, as the droplet contacts the leaf surface (at a high speed since it is being propelled by a pressurized sprayer), the droplet will flex and then snap back into the round shape. This "flex and snap" action will commonly cause the droplet to bounce off the leaf. Second, if the droplet is retained on the leaf, the waxy cuticle repels it and only a small part of the droplet actually contacts the leaf surface. It is through this small area of contact that the herbicide has to diffuse from the droplet into the leaf, which it does quite slowly. An additional challenge is that the droplet quickly starts to evaporate. If the droplet dries before the herbicide enters the plant, the herbicide will often turn into a crystal on the leaf (think of the white residue left behind when saltwater evaporates). If the herbicide crystallizes, the likelihood that it will ever enter the plant is extremely low. The key is to get the herbicide from the droplet into the leaf as rapidly as possible. If the droplet bounces off, is repelled by the leaf or dries too quickly, an insufficient amount of herbicide will enter the leaf and the weed will survive the treatment.

The addition of a wetter/spreader to the spray mixture greatly changes the spray droplet by lowering the surface tension of the water (the forces that make the water form a round bead) and provides three advantages. First, as the droplet contacts the leaf, the lower surface tension means that the droplet no longer wants to form a round bead; instead of bouncing off the leaf, the droplet flattens out and spray retention is greatly improved. Second, the flat droplet contacts much more of the leaf than a round droplet. This increased coverage allows better diffusion of the herbicide into the leaf since more surface area is exposed to the herbicide solution. Third, the addition of the surfactant slows down droplet evaporation, giving the herbicide more time to diffuse into the leaf.

One of the most common questions about wetter/spreader adjuvants is which brand is best. This is a difficult question to answer for many reasons, but in general, the best brand is the one you have successfully used for many years. Problems occur when an applicator attempts to buy the least expensive product (which often changes from year to year). The wisest strategy is to find a brand you are comfortable with and use that as much as possible. When trying a new product, start with a small amount and see if it fits your needs. The labels of many aquatic herbicides provide guidance regarding adjuvant selection; in fact, some products require the use of a particular type of adjuvant. However, don't over-spend because doubling or tripling your adjuvant expenses may not be cost effective. Another common question is what rate of wetter/spreader to use. In general, 0.25% v/v (1 quart of product per 100 gallons of spray mix) works great. There can be an advantage to increasing this to 0.5% v/v, but a rate higher than this rarely results in added benefit. Lastly, not all adjuvants are labeled for application in aquatic environments. Before applying any product to an aquatic system, check the label and make sure the product can be used in or around aquatic sites. The adjuvant label will specifically state "*Not for use in aquatic sites*" if the product cannot be used in, on or over water.

Organosilicones

Organosilicones are a distinct class of spray adjuvants. Their performance is similar to the wetter/spreaders, but organosilicones dramatically reduce (or totally remove) the surface tension forces of water. This causes the droplet to distribute itself into a very thin sheet across the leaf for maximum coverage. Organosilicones work quite well, but they are often more expensive and are not used as often as wetter/spreaders.

Penetrants

Penetrants are oil-based adjuvants and are most often crop oil concentrates and methylated seed oils. Using a water-dispersible oil adjuvant has a clear advantage over a traditional wetter/spreader. Recall that the wetter/spreader does little to improve herbicide uptake beyond ensuring that the droplet lies flat on the leaf. The herbicide must still diffuse through the cuticle to reach the cells and veins below. The waxy cuticle cannot be dissolved by water or a soapy wetter/spreader, but oil *will* soften or dissolve the cuticle. Therefore, as the spray droplet contacts the leaf surface, the oil-based adjuvant begins to dissolve these waxes. As the waxes are stripped away, the herbicide can easily penetrate the leaf and be transported to the regions where it can be most effective.

Since these adjuvants help the herbicide penetrate into the leaf, weed control is often greater with an oil-based penetrant than with a wetter/spreader. Penetrants are typically used on weeds that are larger and more difficult to control, or on species with leaves that are particularly waxy (think waterhyacinth – Section 2.11). Penetrants can also be useful if the weather has been dry, because plant cuticles may thicken to reduce drought stress. If weed control must be performed during these times, an oil-based adjuvant may be essential to help dissolve these thick leaf waxes and facilitate herbicide uptake. You should take into consideration that penetrants are usually applied at a 1% v/v (1 gallon per 100 gallons of spray mix), while wetter/spreaders are added at 0.25% v/v.

It is important to note that penetrant adjuvants are not always the best solution. For example, glyphosate does not perform as well when oil-based adjuvants are used. Conversely, other herbicides should only be used with penetrant adjuvants. It is, therefore, important to read the herbicide label so the recommended adjuvant can be used. Also, since oil-based adjuvants strip away leaf waxes, they can injure desirable plants that are not normally affected by the herbicide. For example, 2,4-D is often used to control broadleaf weeds in grass because grasses are not damaged by 2,4-D. However, if 2,4-D is applied with a high rate of an oil-based adjuvant, the penetrant oil can actually burn the desirable grass since the cuticle is eroded and the cells beneath die when exposed to the environment. The grass will recover, but the injury can be unsightly for a period of time.

Utility adjuvants

Utility adjuvants have a very different role and purpose than activator adjuvants. Activator adjuvants actively promote herbicide uptake into the plant by influencing the spray droplet, the plant cuticle or both, but utility adjuvants improve the efficiency of the spray operation. There are many types, brands and blends of utility adjuvants that have value for their specific uses, but their benefit is often situational and may not provide an advantage across all conditions. Therefore, it is important to understand what these products are designed to do so they can be used to maximum effect.

Defoamers

Wetter/spreader adjuvants are commonly added to improve herbicide performance. These adjuvants are soaps, so foaming is common when the tank is refilled. A small amount of defoamer added prior to tank filling can prevent bubble formation and greatly improve the efficiency of the application. Consider the photo shown at left; though a foam-forming adjuvant was used in both beakers, defoamer was only added to the container on the right. Adding defoamer after a large quantity of bubbles has formed requires much more product and time to clear the tank for refilling. It is important to be proactive and add defoamer to the spray tank before adding soapy adjuvants.

Water conditioners

All natural waters contain dissolved minerals, including iron, magnesium, calcium and aluminum, and these minerals can change the properties of water. For example, the amount or type of minerals in water is what makes water from one region of the country taste different from another. The mineral content of water used in a spray tank can affect application because the minerals listed above are all positively charged, while many commonly used herbicides are negatively charged. When these negatively charged herbicides and positively charged minerals are dissolved in a spray tank together, they naturally attract each other like magnets.

This causes problems because herbicides are highly specific and work by binding to exact places on exact enzymes within the plant. Also, they diffuse through plant cuticles in a specific manner. When a herbicide is bound to a mineral such as calcium or a magnesium complex, it may be unable to enter the plant and work properly. If many herbicide molecules are bound to and deactivated by mineral complexes, they will lose their herbicidal activity and the application will be less effective.

Water conditioners were developed to minimize the impact of dissolved minerals on herbicides. One of the most common conditioners is ammonium sulfate [$(NH_4)_2SO_4$]. Ammonium sulfate and other water conditioners bind to minerals that are dissolved in the water, which makes the minerals unavailable to bind to the herbicide and prevents the herbicide from being deactivated. If mineral content is high (especially with aluminum, iron, calcium and magnesium, which are often considered to be most detrimental), it might be useful to add a water conditioner to the tank prior to adding the herbicide.

If all water contains dissolved minerals, do all applications require water conditioners? Not necessarily; it depends on how high the mineral concentration is in the mix water and how many herbicide molecules could be deactivated. In general, the higher the mineral concentration in the water, the greater the likelihood of herbicide deactivation, and the more likely the need to use a conditioner.

Things to consider:

- The addition of a water conditioner may not always be needed because not all aquatic herbicides are affected by water hardness, so consult the label. If herbicide efficacy is lower than expected, send a water sample to a lab for analysis. If the results say your water is “hard” or “extremely hard”, consider adding a water conditioner.
- If you are using a dry ammonium sulfate product, be sure to use “spray grade”. If not, you may have difficulty getting the product to fully dissolve in water. Spray grade or liquid ammonium sulfate products avoid this problem.
- Add the water conditioner to the tank before the herbicide. Fill the tank 25% full, add the water conditioner, fill to 50%, add the herbicide and fill to 100%.
- Always check the herbicide label before adding a water conditioner. Some labels specifically state that NO ammonium sulfate may be used in the application. Remember, the label is the law.

pH buffers

It can also be important to know the pH of the water used in a tank mix. pH is measured on a scale of 0 to 14 and describes water as acidic (pH 0 to 6.9) or alkaline (pH 7.1 to 14). We often think water is neutral (pH 7), but that is rarely the case. For example, if you live in an area with limestone in the soil, your water pH may be 8.0 or higher. Water pH is important because acidic or alkaline water can react with herbicide molecules, which can affect efficacy. The majority of herbicides we currently use are classified as “weak acids” and they perform better in an environment that is slightly acidic – ideally, water with a pH of 4.5 to 6.5. Therefore, mixing a weakly acid herbicide in alkaline water with a pH of 8 could cause the herbicide to begin to degrade and become less effective.

Does this mean that spray water must always be acidified? Not necessarily. Although herbicide breakdown in the tank can occur if the water pH isn’t correct, this may never be an issue if you mix and spray quickly. Regardless, read the product label to determine whether acidification of tank water is necessary. Some labels recommend that herbicides be diluted with water that has a pH of 6 to 8, while others recommend water with a pH of 4 to 7. If water pH is in the recommended range, no action may be required. However, pH testing can be very useful if you are attempting to optimize your spray program.

Spray dyes

Spot-spray applications can be a highly efficient, selective and cost effective way to manage sporadic populations of unwanted plants. However, these plants are often randomly distributed across a landscape, which complicates spot-spraying. Invariably, some patches will be treated twice, while others are missed entirely. If you plan to perform spot-spray treatments, a non-toxic dye can be added to the spray mix to ensure that each and every weed is treated once. A spray dye is a colorant that stains the weeds that have been sprayed. This gives the applicator an immediate visual cue that a particular weed has been sprayed, or missed. Many different brands and colors of spray dye are currently available, but blue is the most common. The color fades and is gone within 1 to 5 days after spraying.

Drift reducers

Herbicides are a powerful and useful tool to manage unwanted plants while preserving and encouraging growth of desirable species. However, a constant concern is damage to desirable plants that occurs when the herbicide spray drifts, or is blown outside the treatment area. Therefore, care should be taken to avoid or minimize herbicide drift. Sprayers work by pressurizing the herbicide solution and forcing it through a hose to a spray nozzle. When the liquid solution strikes the specially designed nozzle, it fragments (or shears) into individual droplets. For example, note the small, drifting droplets being formed during the high-pressure herbicide treatment shown here. Nozzle type and sprayer pressure affect droplet formation and work together to form large or small droplets. Small droplets are of the greatest concern because they are easily moved by wind currents. One way to manage the proportion of small droplets formed is to include a drift-reducing agent in the herbicide mixture, which will “thicken” the spray solution. Thicker liquids resist shearing into small droplets, so fewer small droplets are formed and the risk of drift is reduced.

Though drift reducers can be quite effective, other techniques should also be employed to manage drift.

1. Spray at the lowest pressure possible. As pressure in the sprayer increases, more small droplets are formed.
2. Avoid spraying in high wind. The higher the wind speed, the more likely droplets will drift. Also, high wind can carry small droplets exceptionally long distances.
3. Avoid spraying into the air when possible. It is often necessary to spray into the air when undesirable trees must be managed. However, spraying in this manner increases the likelihood that droplets will drift.
4. Pay close attention to your surroundings. If valuable or highly sensitive plants are nearby (for example, gardens), closely examine what and where you are spraying and evaluate the likelihood of drift occurring.

5. Although most herbicide drift issues arise from physical movement of spray droplets, some herbicides can turn into a gas and drift as a vapor, particularly on very hot days. This is most common with herbicides such as 2,4-D and triclopyr. Products that are especially prone to drift will provide this information on the label, along with guidelines and requirements to reduce the occurrence of drift.

Summary

Adjuvants are not herbicides and do not directly control unwanted plants, but they work with herbicides to greatly improve efficacy and productivity of herbicide applications. With that in mind, here are a few things to keep in mind when considering the use of an adjuvant:

- Before making an application, ensure that the target weed will be adequately controlled by the selected herbicide. Read the herbicide label and note the appropriate plant size and application timing for the target weed. If the wrong herbicide is chosen or applied in an inappropriate manner, the addition of an adjuvant will rarely improve control.

- Be aware that some adjuvants are blends of several products. For example, it is possible to buy products that adjust pH and act as a wetter/spreader. Before you purchase a blend, make sure all of the components are necessary for the application. Using a blended product is not likely to decrease herbicidal activity, but it can result in an unnecessary increase in cost.

- Some manufacturers suggest that a particular adjuvant is so effective that the application rate of the herbicide can be reduced. Caution should be exercised before reducing a recommended herbicide use rate. Herbicide labels are written after a great amount of data is collected over several years at many locations, so recommended label rates and application methods are time proven. Expecting an adjuvant to do the work of a herbicide can result in reduced efficacy, and more often than not, an applicator is better off following the herbicide label recommendations.

- It has been suggested that the addition of common dish soap or fuel oils (such as diesel) to the spray tank may be equally effective as proper spray adjuvants. This is simply not true. Spray adjuvants have been specifically formulated to enhance herbicide performance without significantly damaging the plant. Adding soaps or fuel oils can disrupt leaf tissue, result in significant foaming and increase expenses, while potentially decreasing herbicide activity. An adjuvant that is specifically designed for the particular application should always be used instead of common household products.

Adjuvant technology has improved dramatically over the past 50 years and many of these products are highly reliable and effective. However, reading all product labels is essential to ensure that all treatment components are used for maximum effectiveness in order to improve the efficacy of any weed management program.

Photo and illustration credits:

Page 185: Cross section of a leaf on a typical terrestrial or emergent plant. Modified from an image by Ninghui Shi; used with permission
Page 186: Water on a lotus (*Nelumbo lutea*) leaf with (left) and without (right) a surfactant; Lyn Gettys, University of Florida
Page 187: Beakers with and without defoamer; Jason Ferrell, University of Florida
Page 189 upper: Using a dye while spot-spraying; Thomas D. Brock, University of Wisconsin-Madison
Page 189 lower: Spray drift from a high-pressure herbicide treatment performed at a distance from the target; Ken Langeland, University of Florida

3.7.4 Aquatic Herbicide Application Methods

William T. Haller: University of Florida, Gainesville FL (emeritus); whaller@ufl.edu

Introduction

All pesticide labels contain very specific information regarding how they are to be stored, handled and applied. It is illegal to use any herbicide in, on or over water unless it is registered by the United States Environmental Protection Agency (EPA) for that purpose and has aquatic use directions on the label (Section 3.7). States may have pesticide use regulations that are more strict than federal regulations; thus, several states require that aquatic pesticide applicators be certified and licensed before they may purchase, handle and apply pesticides and that permits are obtained before aquatic pesticides are applied. Potential users of pesticides should contact state agencies such as county cooperative extension offices, state game and fish agencies or state environmental authorities to ensure compliance with any additional state-specific use restrictions.

A few herbicides may be applied directly from the container; for example, the labels of some copper sulfate herbicides suggest placing the dry granules in a cloth bag and towing the filled bag behind a boat to ensure uniform application throughout the water column. However, the majority of aquatic herbicides must be diluted or mixed with water before application. The purpose of the diluent (water) is to ensure consistent coverage of the target weeds so the herbicide can be absorbed into the plants. Most herbicide labels state that applicators should "use sufficient diluent to obtain uniform coverage of the target weed." Some labels are more restrictive and specify the amount of diluent to be used during application of the herbicide. For example, a label may specify "apply in 50 to 150 gallons of water per acre for adequate coverage". The public often believes that the mixture being applied to weeds is concentrated herbicide, but this is rarely – if ever – the case because herbicides are mixed with large volumes of water. Applicators are required by law to have the label at the application site and it is critical that they read the label carefully before aquatic herbicides are diluted, mixed and applied to ensure that the herbicide is applied in a legal, safe and effective manner.

All pesticides are most hazardous to the applicator when they are in concentrated form and being mixed with diluent (water) to prepare for application. Applicators also may handle the herbicide concentrate several times daily and possibly several days each week. Each herbicide label specifies the type of safety equipment applicators are required to wear to minimize their contact with the concentrate. Thus, although many aquatic herbicides do not have swimming restrictions following treatment, the applicators may be required to wear hazmat suits, gloves and face shields when mixing and applying a product, implying a greater hazard than the treated water actually poses to the public. The concentrate may contain several thousand parts per million (ppm) in the formulation, but the final strength is only 1 or 2 ppm or less when applied and mixed into submersed weed treatment site.

Foliar applications

Foliar herbicides are mixed with water and sprayed on the foliage of floating or emergent plants in a given area. The goal during foliar application of an aquatic herbicide is to obtain good coverage and ensure that the maximum amount of herbicide is taken up by the target weed. Most floating and emergent plants have a waxy layer (cuticle) on their leaves and stems that must be penetrated in order for the herbicide to be taken up by the plant. The labels of some aquatic herbicides suggest or require the addition of surfactants (Section 3.7.3) that dissolve the cuticle and facilitate uptake of the herbicide by the plant. For example, a label may state that "a surfactant may be applied at a rate of 0.25 to 0.5% (1 to 2 quarts per 100 gallons) with the tank mix to get best results". In this example, the addition of a surfactant is not required by the label ("may be used"), so its use is optional; other herbicide labels require ("must be used") the use of surfactants.

Just as carpenters and electricians have specialized equipment for their work, aquatic applicators often have tank- and pump-equipped boats and trucks for the application of herbicide treatments. A typical boat may hold a pump (calibrated to apply from 4 to 10 gallons per minute of a herbicide mix) and a 50- to 100-gallon mix tank. This equipment is calibrated by the applicator to apply the correct amount of herbicide over the area to be treated. Selectivity, or the ability to control weeds growing among native plants, is usually accomplished by choosing the appropriate herbicide or by using a handgun for targeted application of the herbicide mix only to the weeds and not to the desired native species. This is not always possible but is practiced as much as equipment and herbicide selection allow. Riparian owners and lake users may sometimes see damaged or brown native plants in addition to the target weeds 1 or 2 weeks after an herbicide treatment and conclude that all vegetation – including desirable native plants – is dead. However, some non-target plants (particularly perennial and emergent species) often recover and will recolonize the treated site a few weeks following application.

Most homeowners have small "pump-up" garden sprayers or backpack sprayers for lawn and garden use. Herbicide labels may include use directions for mixing the herbicide for small or localized spot treatments using small equipment. For example, if control of clumps of purple loosestrife along a shoreline is desired, the herbicide label may state "mix a 1 to 2% solution of herbicide in a backpack sprayer and spray weeds to wet". A gallon of water contains 128 fluid ounces, so the applicator would add 1.28 fluid ounces of herbicide to 127 fluid ounces of water to get a 1% solution. A 2% herbicide solution would be 2 x 1.28 fluid ounces, or 2.5 fluid ounces of herbicide per gallon of total tank mix. Be careful; some herbicides cannot be used in sprayers that will also be used for garden or ornamental plants, as some leftover herbicides can be quite toxic to other plants. Where is this information? On the label that is attached to every herbicide container!

The foliar application of herbicides to emergent and floating-leaved plants is generally well understood by homeowners because this is common practice for applying insecticides and other products to ornamental, lawn and garden plants. The application of herbicides for submersed weed control, however, is often more complicated and thus more difficult to understand.

Submersed aquatic applications

The control of submersed aquatic weeds is much more difficult than control of emergent aquatic plants for the following reasons:

- Fewer herbicides are registered for submersed treatments
- The amount of herbicide needed depends on the depth of the water
- Wind, waves, inflow, outflow and currents dilute herbicides
- It takes more time to treat and cover submersed plants
- Submersed weeds are generally much more expensive to treat
- The growth stage and area covered by the plants are important
- Use of treated water for irrigation and drinking may be restricted

These general factors – and additional site-specific ones – determine which herbicides should be used to control submersed aquatic weeds. Water flow, dilution and water use are often the critical factors to consider when choosing an herbicide. Water flow and dilution may result in herbicide concentration/exposure times (CET) that are insufficient for herbicides to be effective (Section 3.7.1). There are also water restrictions on many herbicides for use near potable water intakes and water used for irrigation. Herbicide labels will include specific use restrictions.

From the information above, it should be clear that the least complicated, easiest scenario where aquatic herbicides are used to control submersed weeds is in a small pond with no water flow or water use restrictions. There are several herbicides to choose from, and once applied at recommended concentrations, there are no concentration:exposure time or water use concerns. In contrast, a narrow strip of submersed weeds along the shoreline of a 300-acre lake may have the same area and volume as that small pond, but is subject to wind and wave action, water currents and potential water use for irrigation of agricultural crops; therefore, managing this strip is much more complicated and may not be possible.

There are three general types of submersed aquatic weed herbicides based upon their concentration:exposure times and modes of action: contact herbicides, slow-acting systemic herbicides and fast-acting systemic herbicides.

Contact herbicides

Contact herbicides are applied at relatively high concentrations, have very short half-lives in water and require a contact time of hours to a few days to kill plants. They include copper products, diquat, endothall, carfentrazone and flumioxazin which may be applied along strips of shoreline and in relatively small areas where dilution is high, provided contact of the herbicide with the target weed is maintained for an amount of time sufficient to achieve control. The decision to use a contact herbicide is site-specific and the greatest chance of success occurs when herbicide applications are done on calm days to optimize contact times. Contact herbicides in general provide 3 to 6 months of weed control, depending upon the weed, geographical area of application (northern US vs. southern US) and length of growing season.

Systemic or enzyme-inhibiting herbicides

Systemic enzyme-inhibiting herbicides are generally applied at concentrations lower than contact herbicides, must remain in contact with target weeds for relatively long times (up to 45 days or more) and are very slow to control submersed aquatic weeds. These herbicides are often applied as low-dose whole-lake treatments to control weeds throughout the pond or lake. Systemic enzyme-inhibiting herbicides include fluridone, penoxsulam, bispyribac and topramezone. These herbicides are applied at rates of 15 to 45 ppb (parts per billion); concentrations can be maintained with additional treatments over several weeks to control hydrilla (Section 2.2), Eurasian watermilfoil (Section 2.3) and other submersed species. Imazamox is applied at higher rates (100 to 200 ppb) and requires a slightly shorter contact time.

Systemic herbicides with short contact times

There are always exceptions to the rule and the auxin herbicides 2,4-D and triclopyr are the exceptions in this case. Both are systemic herbicides but are absorbed in lethal doses by the target weeds in a relatively short time (1 to 4 days) depending upon the concentration applied. These two herbicides are effective for selective control of Eurasian watermilfoil and other dicot (broadleaf) weeds. Concentrations of these herbicides for submersed weed control generally range from 1 to 2 ppm (parts per million). 2,4-D and triclopyr are applied at the highest labeled dose in areas where dilution is most likely to occur (such as small treatment areas and in strip treatments along shorelines) and on dense mature plants. Lower doses may be used in large treatment areas and in protected coves and bays with little water exchange. A new auxin-mimic herbicide, florpyrauxifen-benzyl, has a concentration:exposure time requirement of as little as a few hours exposure to 5 to 45 ppb depending upon the weed being treated. Most auxin herbicides control broadleaf weeds (dicots), but this herbicide also controls some grass (monocot) weeds as well. Rapid uptake of florpyrauxifen-benzyl and its selective control of many submersed weeds indicates that it can be used in small plot or partial lake treatments. Extensive field research is currently in progress to further delineate the potential of this new product on submersed as well as floating and emergent species.

Application of formulations

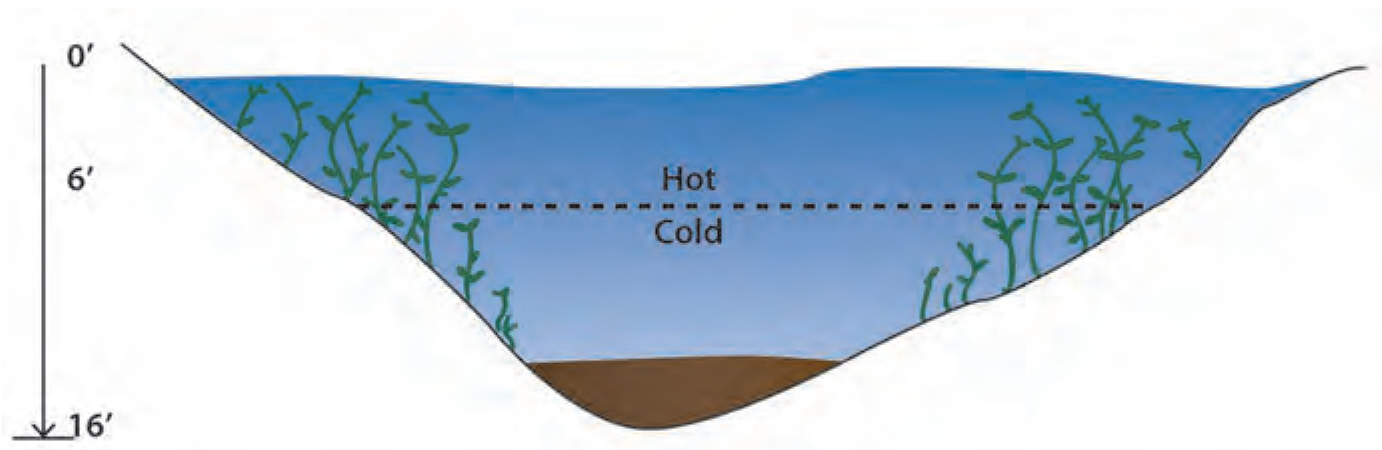
Herbicide formulation refers to how a herbicide is sold (as a liquid, granular or other form) and this determines the type of equipment needed for application of the herbicide. Many aquatic herbicides are sold as both liquid and granular formulations because many are used for both foliar and submersed aquatic weeds. For example, you would not apply

2,4-D as a granular formulation for foliar applications to purple loosestrife (Section 2.16); you would use a liquid formulation. The formulations of aquatic herbicides are listed in Section 3.7.1.

Liquid formulations can be applied to submersed aquatic weeds in several ways, with the type of application determined by the specific location, size and depth of the treatment area. Surface applications are typically done along shorelines and under or around boathouses and docks where water depths average 3 to 6 feet deep. Granular and deep-hose applications are often used in deeper water, particularly in water where submersed weeds are growing in water from 6 to 20 feet deep. The objective of these deep-water treatments is to ensure that the herbicide mixes in the water column and reaches the plant beds where they can be taken up by the target weeds.

Effect of thermoclines

Temperature-dependent thermoclines often develop in lakes and other non-flowing waters during summer, particularly in northern regions. A thermocline occurs when the upper and lower portions of the water separate into warm and cool layers. Swimmers are often familiar with this phenomenon; for example, water in the upper layer of a lake feels warm, but diving down to depths of 6, 8 or 12 feet can be shockingly cold. This thermal stratification is well-known to applicators of aquatic herbicides as well and can reduce the effectiveness of herbicide treatments because the warm upper and cool lower layers of the water do not mix. Herbicides applied to the surface of the water may control upper portions of weeds, but herbicides do not diffuse or penetrate into the deeper cool layers. As a result, root crowns, rhizomes and low-growing plants below the thermocline are not controlled by the herbicide. The depth of the thermocline is influenced by water clarity and varies among lakes, but water temperature within the thermocline typically drops 2 °F for each 3 feet change in depth. If aquatic weeds are growing above and below the thermocline, deep-water injection of liquid herbicides or application of granular herbicides may be used to control weeds in both thermal zones.



Foliar and submersed concentrations

The labels of most aquatic herbicides allow foliar applications for floating and emergent weed problems and the directions for this use are clearly stated on the herbicide labels. Foliar-applied herbicides such as 2,4-D, triclopyr, glyphosate, diquat, endothall, imazapyr and imazamox are usually mixed with 50 to 100 gallons of water per acre treated according to label directions and a surfactant is usually added to the tank mix to facilitate herbicide absorption or to ensure even coverage of the target plants. These herbicides are typically applied in "pounds per acre" with one pound of the herbicide's active ingredient in 100 gallons of water, resulting in a 0.1% concentration (1000 ppm) in the mix tank. This relatively high concentration is needed to ensure that the emergent plant absorbs enough herbicide to kill the weed on contact or through translocation to the site where the herbicide kills the plant. More recently registered systemic enzyme-inhibiting herbicides such as carfentrazone, flumioxazin, penoxsulam, bispyribac and topramezone and the auxin mimic florpyrauxifen-benzyl are applied at much lower rates of only a very few ounces per acre.

Fortunately, the use of herbicides to control submersed aquatic weeds usually requires much lower concentrations of herbicides in the water to be effective. This is because most submersed plants lack the waxy cuticles that slow herbicide uptake in emergent plants and the leaves of many submersed plants are only a few cells thick. Tank mixes may still call for one pound of herbicide in 100 gallons of water, but when diluted in one acre-foot of water, the concentration of herbicide that contacts submersed plants is only 1/2.7 or 0.370 ppm (370 ppb) due to the dilution effect of the water being treated (see Section 3.7.1 for instructions on calculating ppm and ppb). Eurasian watermilfoil can be controlled with as little as 10 ppb of fluridone, but control of this weed with triclopyr or 2,4–D may require up to 2 ppm (2000 ppb). The ability of herbicides to control submersed weeds at such low concentrations contrasts sharply with the concentrations required to control larger, more tolerant floating and emergent weeds. Of course, if the treatment site is very deep (such as 10 to 12 feet), the total amount of herbicide used per acre for submersed weed control increases as well and can exceed the amounts applied in foliar applications, but the final diluted herbicide concentration in the water for submersed treatments is typically less than that contained in foliar sprays.

Selectivity

Weed control in an aquatic ecosystem is very different from weed control in an agricultural setting. For example, farmers want to control all the weeds in a cornfield without affecting the corn, whereas managers of natural and aquatic areas often wish to control a single weed species growing among 50 to 100 desirable native species. Research regarding selectivity of aquatic herbicides is ongoing and depends upon the following factors:

- <u>Choice of herbicide:</u> some herbicides control submersed weeds without affecting a number of other desirable nontarget plants, but the choice of herbicides that work in this manner is limited and complete selectivity is not always possible. As a result, herbicide selection is often dictated by the types of native species present in the proposed treatment area. In general, herbicides applied for submersed weed control have little effect on rooted emergent species due to the relatively low concentrations of herbicides used to control submersed weeds. The use of the water from the proposed aquatic weed treatment site also dictates and plays a significant role in selection of which herbicide to use. The EPA and the Food and Drug Administration (FDA) determine if any potable water use restrictions, crop irrigation restrictions and fishing or swimming restrictions may be necessary to protect human health and the environment. These restrictions are clearly stated on the herbicide labels. Currently, there are no fishing (fish consumption) or swimming/ recreational use restrictions on any of the aquatic herbicides listed in this manual, but irrigation restrictions are more common. Labels for triclopyr and imazapyr state that treated water may not be used for irrigation for 120 days following treatment unless a chemical assay shows less than 1 ppb in the water; diquat has a 5-day irrigation restriction and several other herbicides have irrigation restrictions as well. Always check the label and also your responsible state agency since some states have additional water use restrictions.

- <u>Dose or amount of herbicide:</u> not all plants are equally susceptible to herbicides. Application rates needed to control different weeds are usually listed on the herbicide label.

• Stage of plant growth: some herbicides used for submersed weed control can be applied in very early spring when weeds are actively growing and native plants are still dormant. The use of some herbicides such as glyphosate and imazapyr on emergent perennial grasses such as phragmites (Section 2.17) and cattails seems to be more effective if applied in the late summer or fall when the plants more effectively translocate the herbicide down into the plant roots and rhizomes prior to winter dormancy.

• Selective foliar application: handguns can be used to target and apply herbicides only to the weeds and minimize damage to nontarget species. However, this method is not feasible in most submersed treatments.

Although selective treatment of submersed weeds is more difficult than treatment of floating and emergent weeds, the reduction in growth and coverage of submersed weeds generally results in less weed competition and quick recovery of native species in the treated area. This occurs because most submersed weeds reproduce by vegetative means and many nontarget native plants reproduce by seeds. Elimination of dense weed canopies and the reduction of competition from invasive weeds often results in germination and growth of desirable species during the season of the herbicide treatment or soon thereafter.

Summary

Small-scale foliar application of herbicides to emergent and floating weeds is easily within the capabilities of most riparian homeowners, provided the correct herbicide is chosen and label directions are followed. The application of herbicides to aquatic weeds in large areas or for submersed weed control is more expensive, complicated and often requires specialized equipment to obtain the most cost-effective control. Selectivity results from a combination of factors, including herbicide choice, time of year and nontarget desirable species in the proposed treatment area. The size or area of the treatment site also affects the concentration-exposure time requirements for herbicides. In addition to label requirements, all these factors that affect submersed weed control clearly indicate that experienced state agencies responsible for permitting and managing aquatic resources be contacted prior to undertaking weed control projects.

The discussion in this Section is directed towards riparian homeowner associations and others conducting similar large-scale weed control programs in pubic waters; however, the principles, rules and regulations also apply to the thousands of private pond owners in the US. Aquatic weed control in small ponds is not as complicated as large public operations, but there are some factors to be considered. These include: What is the primary use of the water? Is it used for irrigation? Does water flow from the pond? Are there irrigation or potable water uses downstream? Despite being “private”, some states require permits be obtained to use herbicides in these ponds and may have additional regulations if there is water flow out of the ponds. Aquatic herbicides are usually available in the marketplace in smaller containers or as diluted products for the do-it-yourself pond owner. Also, every state has commercial companies that offer pond management services such as fish stocking, water quality monitoring, aeration, mechanical, chemical and other related services. The Cooperative Extension Service in your county, as well as your environmental state agency, can usually help with plant identification, pond management questions and are likely aware of commercial companies serving your area.

Photo and illustration credits:

Page 191: Herbicide application; William Haller, University of Florida
Page 192: Herbicide application; William Haller, University of Florida
Page 194 upper: Submersed herbicide application with trailing hoses; Thomas McNabb, Clean Lakes Inc.
Page 194 lower: Thermocline; Joshua Huey, University of Florida
Page 195: Herbicide application; William Haller, University of Florida

3.8 A Discussion to Address Your Concerns: Will Herbicides Hurt Me or My Lake?

Bernalyn D. McGaughey: Compliance Services International, Lakewood WA; bmcgaughey@complianceservices.com

1. Our lake is pristine and we don't want to put dangerous chemicals in it. Why should we use herbicides now?

A pristine lake is balanced, stable… and very rare, especially when lakes are surrounded by homes or used for recreation. The lakes we live near and play in are often inundated by excess nutrients and foreign and invasive species. Most water bodies that require herbicide treatment have experienced explosive growth of invasive aquatic plants. While your lake may seem natural and pristine, there are sufficient nutrients in the water to allow unwanted invasive weeds to dominate the system. Control of these weeds will enhance plant diversity and water quality (both of which are degraded by the dense growth common of invasive plants) and will help restore the overall health of the lake.

Hesitancy to utilize aquatic herbicides almost always results in a cycle of trying other methods that are less effective in weed control or more disruptive to the lake system. By the time lake managers turn to the use of herbicides, aquatic weed control is required for a larger part of the lake because weeds have become more dense or infested additional portions of the lake system. In some cases, this delay can be harmful to fish, desirable plants and water uses. In the case of cyanobacterial (blue-green algae) control (Section 2.18), lack of quick action can present risk to drinking water supplies or pets and wildlife drinking from infested waters.

Your lake association or responsible public agency has evaluated all the options for aquatic plant management and has decided that the most effective means of controlling weeds at this point is to use herbicides. The herbicides that will be used are biodegradable or naturally recycled and will not affect the pristine nature of the lake in the long term. When used by professionals according to label directions, herbicides are not "dangerous chemicals" but instead are curative products that have been extensively tested and can effectively control nuisance and invasive aquatic weeds.

2. How dangerous are these chemicals? How do we know they're safe?

Interestingly, aquatic herbicides are one of the smallest niches of specialty weed control products (Section 3.7.1), yet they are also among the most extensively researched and tested. Because these products are added directly to water, the US Environmental Protection Agency (EPA) requires extensive data to assess the safety of a herbicide before it can be registered for use in aquatic systems (Section 3.7). Many years of testing and use have shown that registered aquatic weed control products can be used safely in all areas of the US. In addition, many years of safety and monitoring tests in the laboratory and in the field have been conducted to determine exactly how a given product should be used in a particular situation. It is also important to remember that the treatment level (or concentration in water) of a herbicide is typically much lower (100- to 1000-fold more dilute) than any of the test levels used in laboratory studies that are required to evaluate the safety of the product.

The data required by the EPA for registration of an aquatic herbicide are generated in studies that are conducted according to stringent protocols of conduct, design and evaluation. For example, a given study must be conducted using an EPA approved testing protocol that describes the number of organisms that must be tested, how they are housed and even the temperature and daylength under which the organisms must be maintained. The test is also governed by a series of "Standard Operating Procedures" that have additional parameters for testing and documentation and ensure that the studies are conducted under legally enforceable Good Laboratory Practices by a research facility that is qualified for conducting such tests. Good Laboratory Practice Standards are validated through both internal and external audits. The guidelines for the test are further supported by a "Standard Evaluation Procedure", which outlines for the EPA study reviewer the criteria that must be met in order for the study to be defined as "acceptable." The EPA scientist produces a "data evaluation record" for the study and ultimately classifies the study as acceptable or unacceptable for incorporation into the risk assessment process. Once a study has conformed to all of the requirements for study conduct and acceptance, data generated by the study are combined with data from all other acceptable studies of the herbicide and a risk assessment profile is developed.

The risk assessment process is complex and requires identifying which studies should be integrated into the hazard and exposure evaluation process. The 84 to 124 different studies required for registration of an aquatic herbicide take from 6 to 10 years to complete and are integrated in a robust scientific assessment that is evaluated by the EPA in a process that can take an additional one to three years before labels are approved. Often state or local regulatory agencies require additional, site specific assessment of the herbicide to further validate that local conditions and use patterns do not present non-target organism or human risk.

In addition to the review and risk assessment conducted when a product is first registered, EPA has a registration review cycle during which every registered pesticide must be reevaluated. This entails not only a look-back at previously reviewed and relied-upon data, but also the assessment of products using new data, published literature, new modeling techniques, and a review of any incidents that may have been reported in connection with the use of the product.

3. Do these herbicides break down in the environment? I realize the herbicides themselves have been evaluated by regulatory agencies but what about their breakdown products?

Identification and evaluation of the components into which a herbicide breaks down is a critical and required part of the data that must be submitted as part of a product's registration process. Degradation and metabolism pathways must be studied and the molecules that are produced along those pathways must be identified. If any molecules are believed to be "of toxicological concern" (and there is a definition for that), then those molecules must be tested as well, both alone and in combination with the original or "parent" molecule.

Testing of breakdown products is not limited simply to toxicity; breakdown products must also be evaluated for their persistence in the environment. In addition, the mechanism (light, heat, microbial action) that produces them and acts to further break them down must also be understood. The final fate of the parent and breakdown products must be completely identified, reported and understood by chemists and toxicologists. Additionally, there are regulatory flagging criteria that are used to put "stop lights" on certain uses or environmental introductions of herbicides. These "stop lights" can be associated with direct toxicity, persistence, bioaccumulation or other important environmental and toxicological properties of the pesticide. If a product is flagged by one of these "stop lights" during testing, the company developing the product (especially one that will be used in water) may reconsider whether to proceed with the high cost of registration if there is a good chance the product will not successfully make it through the registration process.

4. If the chemical companies do the research and submit their data to the EPA, isn't this like the fox guarding the henhouse? Their data may be falsified!

With the current regulatory standards and rigor of EPA review, it is virtually impossible to falsify the data supporting a product and would be extremely foolish to even attempt. Companies submitting studies must certify that they are conducted in accordance with EPA regulations for good laboratory practices and companies usually hire independent quality assurance scientists to conduct audits as the studies are performed. In addition, the EPA has established a random laboratory and study audit program. This program has the authority to audit laboratories that conduct studies in support of pesticide registration and companies that sponsor them, and can randomly select submitted studies for auditing. It must be possible during this audit process to confidently recreate the entire study from the "raw data" (laboratories are legally required to maintain all data for any submitted study on which registration relies). If a problem is found or the results cannot be reconstructed, not only is the study rejected for regulatory use, but the facility conducting it or the company sponsoring it is likely to undergo a more complete audit of all studies conducted during the same period, at the same facility or on the same product. Penalties for falsifying studies can be severe and include fines and/or imprisonment.

5. If herbicides make up only part of the chemicals that are applied, how do we know whether any other part of the product or its inert ingredients are dangerous?

First of all, let's understand a little bit about herbicide formulations. The chemical that controls the weed, in its pure form, is called the "active ingredient". The "technical grade" of the active ingredient is used in testing, and that technical grade must contain all those components that are found in the typical manufactured product that makes up the active ingredient. Technical grade chemicals are usually very pure (98%+), but may include additional compounds that are formed as the active ingredient is made. Components in the technical grade product, other than the pure active ingredient, are usually remnants of the manufacturing process, molecules that are impossible to separate from the parent compound, or other unintentionally added or formed ingredients. All such impurities must be identified even if they are present in extremely low quantities. If any are of toxicological concern, they must be removed from the technical product or reduced to levels considered acceptable by the EPA.

Testing with the technical grade of the herbicide will identify toxic and environmental effects that might be caused by the active ingredient itself or any chemical components that cannot be removed from the active ingredient. However, the technical grade form of herbicides are too concentrated to be used without additives and are rarely useable as herbicides without some modification to allow proper measurement (dilution by water, clay granules or other solvents or carriers), tank mixing (conditioners, such as emulsifiers, anti-foaming agents or wetting agents), and stability and distribution to the target site (by use of surfactants, drift control agents, dyes or other similar agents) (Section 3.7.3). The proper addition of these materials to the technical grade product produces what is called an “end use formulation,” which is the commercial product used in weed control. This end use formulation must also be tested, but in a limited way unless the initial tests show that there is a measurable difference in toxicity between the technical product and its end use formulation. If there is a difference, the typical remedy is to change the components of the formulation so that they do not affect the toxicity or environmental characteristics of the end-use formulation. Once all EPA requirements are satisfied for the end use formulation, and EPA scientists have reviewed the proposed use of and data on it, then a registration for the end use formulation is issued and the product can be sold.

Collectively, the technical grade by-products and purposely added formulation ingredients discussed above are often referred to as “inert ingredients” because they do not contribute to the activity of the active ingredient. Formulations are considered trade secrets because their components may provide a competitive advantage which will be associated with a brand trademark. As such, the “secrecy” surrounding inert ingredients is one of competition, not toxicological properties. Additionally, not just any compound can be used in a formulation. The EPA requires that all inert ingredients in pesticide products be cleared prior to use in a formulation and the EPA maintains a list of products from which the formulation chemist can choose. If the formulation chemist chooses a product that is not on the cleared list of inert ingredients, then supporting data must also be submitted for that “inert” ingredient. A separate and thorough review process will determine whether the inert ingredient can be added to the EPA’s cleared list and safely used in the subject formulation. Incidentally, these inert ingredients are not “secret” from the EPA. Each technical and end use product must be supported by a complete “confidential statement of formula” so that the EPA can evaluate the acceptability of the full product and its additives. The confidential statement of formula is also used by the EPA when random or purposeful samples of the product are pulled from chemical distributors or applicators and analyzed for their compliance to the stated formula. If the product is found to vary from its stated formula, it is considered misbranded and misbranding comes with its own set of fines and possible imprisonment.

Inert ingredients in products to be used on food (and most aquatic uses are considered food uses due to the subsequent exposure to fish and shellfish, which in turn could be food items for people) or potable water must also have tolerances (allowable exposure levels for the product and any breakdown products of concern) set under the Federal Food, Drug and Cosmetic Act, which is administered by the Food and Drug Administration. Scrutiny of products that are used in, or may reach drinking water sources, is especially intense because the underlying assumption is that exposure could occur over a lifetime, from any and every drinking water source. In the case of aquatic herbicides, this assessment process greatly overstates exposure and thus results in a very conservative risk assessment when EPA reviews these uses for registration.

6. When will it be safe for my kids to swim in the water again?

Each herbicide has a specific label statement regarding water use and swimming after weed treatment. Label statements are based on the results of various studies and the regulatory risk assessment process described above. Swimming restrictions listed on the label are most often related to the dissipation of the herbicide in water and added “safety factors” that build in at least a 100- to 1000-fold margin between what is observed in studies as a “no effect level” and the potential exposure level when a lake is treated. Therefore, the restriction interval (if any) is related to all studies conducted on the degradation and dissipation of the product and its dermal, oral and dietary toxicity, as well as any potential it has to irritate the skin or eyes or penetrate the skin. Herbicides that lack swimming restrictions may dissipate very quickly and/or the toxicity of the product at treatment levels is far below the “no effect level” in studies supporting product registration.

7. Will herbicide treatments kill the fish in our lake?

Aquatic herbicides are extensively tested for their effects on fish and other nontarget aquatic organisms. For the most part, these products are relatively non-toxic to fish because their mode of action (the way they affect the target weed) is based on photosynthesis or other plant processes that differ from animal biochemistry. A few types of aquatic herbicides (usually algicides) are toxic to fish at or near treatment levels, but application techniques that provide fish with the opportunity to escape from treated waters can reduce or prevent the loss of fish populations. This information is on the herbicide label; the “label is the law”, and applicators are required to read and follow all label directions and precautions.

The applicator must consider the amount of plant cover and the manner in which it will be treated in their professional assessment of the needs of the lake. Decomposing vegetation can deplete oxygen levels in water, which can cause fish mortality or algae outbreaks if application precautions are not taken. Extreme infestations of weeds may require treatment of the lake in stages instead of using a single whole-lake treatment. Partial treatment will allow fish to escape to untreated, oxygenated waters as target plants in the treated area decompose.

8. The herbicide label says that the product is “toxic to fish and wildlife”. Does this mean the herbicide treatment will kill our fish? If not, why do these chemicals kill plants without harming people or fish?

The statement referenced here historically has been required on a label when a pesticide intended for outdoor use contains an active ingredient with a fish LC50 (acute toxicity level) of less than 1 ppm [equal to one part (or molecule) herbicide per one million parts (or molecules) of water]. “LC50” is an abbreviation for “lethal concentration 50%” and represents the calculated concentration of the substance that is expected to kill 50% of the organisms studied. The standard label statement required in this case is, “This pesticide is toxic to [fish] [fish and aquatic invertebrates] [oysters/shrimp] or [fish, aquatic invertebrates, oysters and shrimp]”. Likewise, if the product “triggers” a toxicity level preset for birds or mammals, a similar statement is required. When a pesticide intended for outdoor use contains an active ingredient which has a mammalian acute oral toxicity of less than 100 mg material/kg body weight, an avian acute oral toxicity of less than 100 mg/kg, or a subacute dietary toxicity of less than 500 ppm (500 parts of material per 1,000,000 parts diet, by weight), the label must state “This pesticide is toxic to [birds] [mammals] or [birds and mammals]”. The same types of parameters are applied to pollinators. It is important to note that pesticides with lower LC50 values are more toxic than those with higher values. For example, a product with a toxicity of 100 mg/kg is more toxic than one with a toxicity of 250 mg/kg. Thus, these standard required warnings on the label are associated with the direct toxicity of the product that is expressed in feeding or exposure studies, and not with the treatment-level concentrations that are applied to the lake.

There are several circumstances that can make toxicity to organisms in the field less severe than suggested by the label statement when herbicides are used for weed treatment. Some of these are:

- <u>Effective control levels</u>: most aquatic herbicides are applied at rates well below those that would cause fish or wildlife toxicity. This is either because the target weed is particularly sensitive to the herbicide or because the herbicide interrupts a biochemical pathway that animals do not possess.

- <u>Application techniques</u>: your professional applicator or supervising state agency knows what precautions to take for products that have a treatment rate close to a wildlife effect level. These precautions can include partial lake treatments; optimal treatment timing at the lowest rate possible; the use of drift control agents; and other informed choices made by the professional applicator.

- <u>Dissipation rate:</u> Some aquatic herbicides break down immediately or are rapidly absorbed by plants and vegetative matter. Studies to determine fish toxicity are conducted in pure-water systems (without plants or sediments) over a period of several days. Such studies provide comparable standards for judging toxicity and regulating products, but they are not necessarily equal to fish exposure and product toxicity in a natural, living system when a herbicide is used according to label directions.

• Sediment binding: Some aquatic herbicides ultimately bind to organic matter, algae and soil particles and partially end up in lake sediments, where they may be metabolized by microbes or made unavailable through the physical process of mineralization. A product that is bound in the soil this way rarely presents a toxicity concern.

9. Is it safe to eat fish from the lake after herbicides have been applied?

No aquatic herbicides currently registered by the EPA have fish consumption restrictions. There are no restrictions because herbicides have established "tolerances" that are set by the EPA and the FDA. Tolerances are boundaries for acceptable levels of pesticide residues in food and are established after review of submitted data and in accordance with the Federal Food, Drug and Cosmetic Act. If an aquatic herbicide has tolerances set for fish, then the label will instruct whether the fish can be consumed immediately after treatment or if there is a waiting period. Where there is no established tolerance (either because the registrant has not sought it or due to the properties of the product), the label will prohibit the consumption of fish from a treated lake until enough time has passed for no residues of the product to be found in fish tissues. Professional applicators are well aware of the restrictions necessary for fishing and fish consumption. These restrictions are clearly specified on the herbicide label. Applicators are required to post signs or otherwise clearly inform lake users of any water use restrictions.

10. How long does it take for herbicides to break down? Do the chemicals become concentrated in the fish or the sediment of the lake?

There are some specialized terms that will help you understand the metabolic processes that are at the root of this question. They are adsorption, depuration, bioaccumulation and bioconcentration. Adsorption is the manner and rate at which an organism or substrate assimilates a chemical into its system, whereas depuration is the manner and rate at which the organism or substrate rids itself of a chemical. Bioaccumulation occurs when the rate of adsorption (taking up the chemical) exceeds the rate of depuration (ridding of the chemical) during the period of exposure. When exposure is stopped, depuration continues and the organism will gradually clear itself of the chemical. Some scientists debate whether there is a difference between bioaccumulation and bioconcentration. However, bioconcentration is slightly different than bioaccumulation because the levels of a chemical that bioconcentrates build up and become more concentrated over time. This occurs because depuration is non-existent or very slow, so the organism never clears the chemical from its system and may build up higher and higher concentrations upon every exposure to that chemical. Bioconcentration does not occur in any currently registered aquatic herbicide. A herbicide may have a short bioaccumulation period in edible organisms like fish and in such a circumstance if an established tolerance would potentially be exceeded, the product would be labeled with restrictions to prevent consumption until the depuration process has cleared the chemical from the organism's system.

Some aquatic herbicides (for example, those containing copper) may accumulate in sediments, but as discussed above, this is typically also associated with sediment binding that limits the biological availability of the product. In the case of copper, which is also a trace nutrient, there are metabolic pathways in sediment systems and plants and animals that assimilate, bind or excrete copper when exposures are at low levels, as they are with aquatic uses of copper. The EPA takes into account potential accumulation of pesticides in fish and sediment prior to registering any product for use in water. In fact, pesticide accumulation in living systems or the environment is one of the "stop lights" discussed in Question 3 above. It is unlikely that any chemical that bioconcentrates would be registered for outdoor use in today's regulatory environment. It is possible that a product that mildly bioaccumulates might be registered, because in most instances this property can be managed by reducing application rates, increasing intervals between treatments and restricting consumption of treated organisms for a given interval until depuration brings the active ingredient below any level of concern. If risks to man or the environment are unacceptable or unmanageable, then the product simply will not be registered.

11. Are aquatic herbicides carcinogens? Will they give me cancer?

There are currently no registered aquatic chemicals that are classified as carcinogens. The treatment of water systems with herbicides is considered a widespread use with high potential for human and nontarget organism exposure. Consequently, products registered for use in water must present a very low risk profile, even when – in the case of aquatic herbicides – potential exposure to humans is neither pervasive nor long term. Any legitimate evidence of carcinogenicity would immediately put the registration and use of an aquatic herbicide in jeopardy.

This brings up an area that confuses many people – how to interpret different kinds of studies with respect to their validity for use in the "risk equation". A number of factors contribute to the validity of a study, such as the purity and reliability of the test system (contaminants not found in the product or nature; the use of unusual species or strains of test animals that could create false results); the statistical power of the experiment itself (inadequate numbers of test organisms or improper statistical analysis of results could yield false conclusions); or the route of exposure (an exposure route impossible in nature, such as intravenous injection of high concentrations of chemical). For these and other reasons, some studies are not used in the risk assessment process, provided there is a body of reliable information that contradicts their findings. In the event a new finding is of concern, the EPA has the means to restrict use, cancel use or put other protective measures in place until additional data are generated or assessed and found to support continued registration and use.

12. Plants that have been treated with herbicides rot and sink to the bottom of the lake and cause a buildup of muck. We don't want muck buildup so we shouldn't use herbicides, right?

The best time to treat with herbicides is usually in the spring when plants are very actively growing but still small. This practice results in very insignificant organic matter additions to the lake. Furthermore, research has shown that when the growth of plants is restricted or controlled with herbicides or other means, much less organic matter is produced than if plants are left untreated. Plants that are not managed in some way grow until they reach their full annual biomass and then naturally die back each winter; as a result, all the material produced by a plant over the course of the year is added to the lake annually. By reducing plant growth, herbicide use can actually reduce organic matter production and accumulation. Another factor contributing to "muck" is sedimentation. Dense stands of weeds tend to trap particles suspended in the water column and increase sedimentation or "muck" buildup.

13. I've watched herbicide applications in other lakes and the applicators always wear "moon suits" and all sorts of protective gear even though the label says we can swim and fish immediately after application of the herbicide. This makes no sense – what gives?

Pesticide labels are developed to take into consideration both the exposure to workers (handlers and applicators) and the exposure to the environment. Workers repeatedly handle concentrated herbicides before they are diluted for application on a daily basis. Therefore, applicators are required to wear specific types of personal protective equipment, depending on the properties of the product, to minimize their exposure to high doses of chemical. Herbicides are diluted literally millions of times when they are applied to water and they are usually applied once per season. As a result, the precautions relevant to an applicator are simply not necessary for any lake water users who are not repeatedly exposed to high concentrations of herbicides. For comparison, a tablespoon of salt in a batch of yeast dough contributes to the flavor and perfection of the final loaves of bread – but a tablespoon of salt taken alone once every day could be dangerous for you.

14. People used to say that DDT, chlordane and all those other pesticides were safe and now they're banned. Will this happen with more modern herbicides too?

DDT was first registered as a pesticide in the 1940s; chlordane was first registered in 1948. Both of these compounds were insecticides and are in no way related to any currently registered aquatic herbicides. There is absolutely no comparison to the testing standards and regulatory requirements in place today with the meager tests that were in place in the first half of the last century. Needless to say, our understanding of science, toxicology and the environment has increased tremendously in the last 80 years.

The oldest registered aquatic herbicide appeared first in the late 1950s. Any products surviving since then have been subjected to additional reviews and many additional data requirements, culminating in updated and more rigorous risk assessments, including continuing registration reevaluations. It is a testimony to their safety that, as testing and registration requirements increase, older aquatic herbicides are still in use today. In fact, with the additional testing, many restrictions have actually been removed from older products. Products developed over the course of the last 30 years, during the continuing development of increased understanding and advanced science, are designed to have a minimal impact on the environment and are simply not comparable to the "first generation" pesticides like DDT and chlordane. Today's products are developed with the knowledge of their toxicity, modes of action and impact, and would not be developed or registered if they carried a high "risk burden".

15. I agree that we have to use herbicides to get our weed problem under control, but how can we as residents reduce the risks associated with the use of these chemicals?

First of all, by taking the time to read and understand this manual, you have already invested in reducing your own risks, because you now understand the importance of following label directions and the instructions provided to you by your professional applicator.

Second, plan carefully and completely for a herbicide application in the early stages of an aquatic weed infestation so that your lake can be treated at the optimum time of the year with the lowest effective treatment rates, which can reduce the need for multiple treatments and the size of the area potentially needing treatment. This action will likely provide more effective weed control, reduce costs and lower the total amount of chemical that may be required for adequate weed control.

Additionally, many states have regulatory agencies that conduct additional risk assessments to refine their understanding of product properties as specifically as possible for the conditions in their state. In some cases, specific permits or precautions are required on a treatment-by-treatment basis, thereby further ensuring that lake residents and users understand the restrictions, if any, on the use of the lake or its resources.

The risk-reducing protections necessary for safe use of a registered product are already in place once the product is registered. As a lake resident, all you have to do to reduce risks is follow the label, the instructions of the applicator and any additional local regulations or postings.

16. What exactly is risk? I don't want any risk!

We cannot live in a risk-free environment. Living near a lake is in itself a "risk". Risk, as related to the science of risk assessment, is poorly understood by anyone other than risk-assessment scientists. Most people equate "risk" with "being exposed to a risk", but these are not the same thing. Risk assessors deal with the likelihood (or probability) of an event happening at all, while being *at risk* is the likelihood of being affected by an event that is known to happen. Thus, the risk assessor will come to a conclusion (for example) that a given dose of a chemical has a one in a million chance of causing cancer, (that is, a probability of one chance in a million that the product might cause cancer), while the statistician following causes of death will report that an individual has approximately a one in four chance of dying from cancer (that is, the cancer rate in the population). Two very different endpoints.

When we put actual quantifiable risks in perspective, the risk of harm from an aquatic herbicide (or any pesticide, for that matter) is negligible. The National Safety Council (2016) reports the following:

- The leading causes of death in the US are heart disease, cancer, preventable injury, respiratory disease and stroke, in that order.

- Of unintentional accidents, the seventh ranked cause of death is drowning. The odds of drowning are 1 in 1,086.

No risk estimate for the effects that might result from exposure to a pesticide even begins to approach this number.

In risk assessment, the end point sought is that the probability of a risk is so low that it is expected to not occur. In risk assessment, "risk" is defined as the relationship between hazard and the likelihood of exposure. When aquatic herbicides are used in a lake, most residents and lake users will have little or no exposure to the product used for weed treatment, based on the application methods, precautions taken and infrequency of treatment. Your risk of suffering from an event related to herbicide use and exposure is miniscule.

17. Does the EPA guarantee that these herbicides are safe?

The regulatory language of FIFRA (Section 3.7) actually prohibits descriptive language that would imply any registered pesticide is "safe". In part, this is because "safe" is a relative term that could easily be misleading. No agent, natural or man-made, is completely "safe". Even water, which is essential for life, can be dangerous if too much is consumed because in excess it can disrupt the balance of electrolytes in a living system. Electrolyte imbalance can lead to shock and eventual death if not corrected.

As discussed above, EPA registration requirements and the risk assessment process supporting a pesticide registration are intense and thorough. The directions for use that are listed on the product label take into account risk management measures that are necessary to reduce the risk of exposure to the point where there is no reasonable expectation of environmental or human health effects. Furthermore, there is now a revolving and formal Registration Review process, as mentioned above, assuring that new scientific procedures and risk assessment methods are applied through a repeated process applied to all EPA registered products at least every 15 years, over the life of their registration.

18. Who else studies these chemicals besides the EPA?

Chemical use and its effects on the environment are closely scrutinized by many groups, including independent university scientists, state regulatory agencies, environmental groups and even the chemical companies themselves. Additionally, as the world economic and regulatory systems become more global, there is a closer coordination between countries in their requirements for and review of data on chemicals.

There are also protections written into FIFRA with respect to the discovery of previously unobserved effects. If a legitimate new, unexpected or adverse result is made known to the company holding the registration for the chemical, that company must, within 15 days, report that finding and its significance to the EPA. Failure to follow these reporting requirements carries heavy penalties. If the EPA deems that the event is critical, it can immediately stop the sale or otherwise limit the use of the product. If the significance of the event is not major, but requires further understanding, the EPA may issue additional data requirements so that the initial finding can be studied and causes for it can be determined. For example, several years ago the possibility that certain pesticides might be endocrine disrupters was of regulatory concern. EPA developed a three-tiered testing program for determining the existence and extent of any adverse effect on endocrine. Many compounds were subject to the first tier of testing, including registered pesticides, and a few to the second. After many years of these initial tests, the testing program was minimized because the condition of "endocrine disruption" by new or currently registered pesticide products was never confirmed.

19. Big corporations are only interested in making money – they don't care whether their product is safe!

The development, registration and marketing of a pesticide take place in a highly visible segment of business in which relatively few companies compete. Add to that the extra burden of registering products for use in water systems and the general business risk couldn't get much higher. This is a mature industry with extremely high standards, a heavy regulatory obligation and a tremendous amount of exposure. Corporations employ scientists to conduct the research required for pesticide regulation, and these scientists eat the same food and use the same resources that we all enjoy. No company in such an environment would survive negligence, data falsification or poor business ethics. The mistakes of the early years that occurred in an emerging regulatory system and a budding scientific understanding of the environment that surrounds us are simply not relevant to the business of today. They are of the past. Today's aquatic herbicide registrants are heavily invested in the safe and beneficial use of their products, environmental stewardship and sustainable practices. They have to be, or they wouldn't be here tomorrow. And being here tomorrow is how they survive, not simply by making money with no future in sight.

For more information

1.1 Impact of Invasive Aquatic Plants on Aquatic Biology

Plant growth form definition: http://www.dnr.state.mn.us/shorelandmgmt/apg/wheregrow.html

Lake food chains: http://www.waterontheweb.org/under/lakeecology/11_foodweb.html

Lake trophic state: https://lakewatch.ifas.ufl.edu/media/lakewatchifasufledu/extension/pamphlets/TrophicState.pdf

Depth zonation of aquatic plants: http://www.niwa.co.nz/news-and-publications/publications/all/wa/10-1/submerged

Invasive aquatic plants: http://plants.ifas.ufl.edu; http://www.dnr.state.mn.us/invasives/aquaticplants/index.html

1.2 Impact of Invasive Aquatic Plants on Fish

Dibble ED, KJ Killgore and SL Harrel. 1996. Assessment of fish-plant interactions. Amer Fisheries Soc Symp 16:357-372.

Etnier DA and WC Starnes. 1993. The fishes of Tennessee. The University of Tennessee Press, Knoxville TN.

Horne A and C Goldman. 1994. Limnology (2nd ed.). McGraw-Hill.

Jeppesen E, M Sondergaard, M Sondergaard and K Christofferson. 1998. The structuring role of submerged macrophytes in lakes. Springer, New York NY.

Kovalenko K, ED Dibble and R Fugi. 2009. Fish feeding in changing habitats: effect of invasive macrophyte control and habitat diversity. Ecol Freshwater Fish 18(2):305-313.

Lee DS, CR Gilbert, CH Hocutt, RE Jenkins, DE McAllister and JR Stauffer, Jr. 1981. Atlas of North American freshwater fishes. Publication #1980-12, North Carolina Biological Survey.

Mettee MF, PE O'Neil and JM Pierson. 1996. Fishes of Alabama and the Mobile basin. Oxmoor House, Inc., Birmingham AL.

Ney JJ. 1978. A synoptic review of yellow perch and walleye biology, p1-12, In: RL Kendall, ed. A symposium on selected coolwater fishes of North America. Amer Fisheries Soc Special Publication 11, 437.

Pflieger WL. 1978. The fishes of Missouri. Missouri Department of Conservation.

Robinson HW and TM Buchanon. Fishes of Arkansas. 1984. The University of Arkansas Press, Fayetteville AR.

Ross ST. 2001. The inland fishes of Mississippi. University Press of Mississippi, Oxford MS.

Schultz R and ED Dibble 2012. Effects of invasive macrophytes on freshwater fish and macroinvertibrate communities; role of invasive plant traits. Hydrobiology 684:1-14.

Scott WB and EJ Crossman. 1973. Freshwater fishes of Canada. Fisheries Breeding of Canada Bulletin 184:1-966.

Webb KM, RE Schultz and ED Dibble 2016. The influence of invasive aquatic plant removal on diets of bluegill in Minnesota lakes. J Aquatic Plant Manage 54:37-45.

1.3 Impact of Invasive Aquatic Plants on Waterfowl

Baldassarre GA and EG Bolen. 1994. Waterfowl ecology and management. John Wiley and Sons, New York.

Havera SP. 1999. Waterfowl of Illinois: status and management. Illinois Natural History Survey Special Publication 21.

Johnson FA and F Montalbano. 1984. Selection of plant communities by wintering waterfowl on Lake Okeechobee, Florida. J Wildlife Manage 48(1):174-178.

Johnson FA and F Montalbano. 1987. Considering water habitat in hydrilla control programs. Wildlife Soc Bulletin 15:466-469.

Kantrud HA. 1986. Effects of vegetation manipulation on breeding waterfowl in prairie wetlands, a literature review. US Fish and Wildlife Service Technical Report 3.

Kantrud HA. 1990. Sago pondweed (*Potamogeton pectinatus* L.): a literature review. US Fish and Wildlife Service Resource Publication 176.

Martin AC and FM Uhler. 1939. Food of game ducks in the United States and Canada. US Department of Agriculture Technical Bulletin 634.

McAtee WL. 1918. Food habits of the mallard ducks of the United States. US Department of Agriculture Technical Bulletin 720.

Perry MC and FM Uhler. 1982. Food habits and distribution of wintering canvasbacks on Chesapeake Bay. US Fish and Wildlife Service, Patuxent Wildlife Research Center.

Smith LM, RL Pederson and RM Kaminski (eds). 1989. Habitat management for migrating and wintering waterfowl in North America. Texas Tech University Press, Lubbock TX.

Soons MB, C van der Vlugt, B van Lith, GW Heil and M Klaassen. 2008. Small seed size increases the potential for dispersal of wetland plants by ducks. J Ecology 96:619-627.

1.4 Impact of Invasive Aquatic Plants on Aquatic Birds

Ehrlich PR, DS Dobkin and D Wheye. 1988. The birders handbook. A field guide to the natural history of North American birds. Simon and Schuster Inc. New York.

Hanson AR and JJ Kerekes. 2006. Limnology and aquatic birds. Proceedings of the fourth conference working group on aquatic birds of Societas Internationalis Limnologiae (SIL). Springer, Dordrecht, The Netherlands.

Kerekes JJ and B Pollard (eds.). 1994. Symposium proceedings: aquatic birds in the trophic web of lakes. Sackville, New Brunswick, Canada. Aug. 19-22, 1991. Developments in Hydrobiology, vol. 96. Reprinted from Hydrobiologia, vol. 279/280.

Peterson RT. 1980. A field guide to the birds east of the Rockies. Fourth edition. Houghton Mifflin Company. Boston.

Terres JK. 1980. The Audubon Society encyclopedia of North American birds. Alfred A. Knopf. New York.

1.5 Aquatic Plants, Mosquitoes and Public Health

Connelly CR, CN Mores, CT Smartt and WJ Tabachnick. 2017. ENY-736: Chikungunya. UF/IFAS Cooperative Extension Service, Gainesville FL. http://edis.ifas.ufl.edu/in696

Dame D and T Fasulo (eds). 2008. Public health pesticide training manual (SP318). University of Florida Cooperative Extension, Gainesville FL.

Johnson LR, JP Cuda and N Burkett-Cadena. 2017. EENY-694: Cattail mosquito (suggested common name) *Coquillettidia perturbans* (Walker) (Insecta: Diptera: Culicidae: Culicinae: Mansoniini). UF/ IFAS Cooperative Extension Service, Gainesville FL. http://edis.ifas.ufl.edu/in1192

Lounibos LP, BW Alto, ND Burkett-Cadena, CC Lord, CT Smartt, CR Connelly and JR Rey. 2016. ENY-888: Zika, a mosquito-transmitted virus. UF/IFAS Cooperative Extension Service, Gainesville FL. http://edis.ifas.ufl.edu/in1120

O'Meara G. 2003. Mosquitoes associated with stormwater detention/retention areas. UF/IFAS Cooperative Extension Service, Gainesville FL. ENY627/ MG338. http://edis.ifas.ufl.edu/MG338

University of Florida/IFAS Florida Medical Entomology Laboratory. Mosquito information website. https://fmel.ifas.ufl.edu/

Wilson F. 1981. The aquatic weed-mosquito control connection. Aquatics Fall 3:6, 9-11, 14.

2.2 Hydrilla

Archambault JM and WG Cope. 2016. Life stage sensitivity of a freshwater snail to herbicides used in invasive aquatic weed control. Freshwater Mollusk Biol and Conservation 19:69-79.

Langeland KA. 1996. *Hydrilla verticillata* (L.f.) Royle (Hydrocharitaceae): the perfect aquatic weed. Castanea 61:293-304. http://plants.ifas.ufl.edu/node/184

Madeira PT, CC Jacono and TK Van. 2000. Monitoring hydrilla using two RAPD procedures and the nonindigenous aquatic species database. J Aquatic Plant Manage 38:33-40. http://apms.org/japm/vol38/v38p33.pdf

McLane WM. 1969. The aquatic plant business in relation to infestations of exotic aquatic plants in Florida waters. J Aquatic Plant Manage 8:48-49. http://apms.org/japm/vol08a/v8p48.pdf

Miller, JL, WT Haller and MS Glenn. 1993. Turion production by dioecious hydrilla in North Florida. J. Aquatic Plant Manage 31:101-105.

2.3 Eurasian Watermilfoil

Grace JB and RG Wetzel. 1978. The production of Eurasian watermilfoil (*Myriophyllum spicatum* L.): A review. J Aquatic Plant Manage 16:1-11. http://www.apms.org/japm/vol16/v16p1.pdf

Madsen JD. 2005. Eurasian watermilfoil invasions and management across the United States. Currents: The J Marine Education 21(2):21-26.

Pfingsten IA, L Berent, CC Jacono and MM Richerson. 2020. *Myriophyllum spicatum* L. US. Geological Survey, Nonindigenous Aquatic Species Database, Gainesville, FL, https://nas.er.usgs.gov/queries/FactSheet.aspx?speciesID=237

Smith CS and JW Barko. 1990. The ecology of Eurasian watermilfoil. J Aquatic Plant Manage 28:55-64. http://www.apms.org/japm/vol28/v28p55.pdf

2.4 Curlyleaf Pondweed

Minnesota Department of Natural Resources. http://www.dnr.state.mn.us/aquatic_plants/submerged_plants/curlyleaf_pondweed.html

Netherland MD, JD Skogerboe, CS Owens and JD Madsen. 2000. Influence of water temperature on efficacy of diquat and endothall versus curlyleaf pondweed. J Aquatic Plant Manage 38:25-32. http:// www.apms.org/japm/vol38/v38p25.pdf

University of Florida Center for Aquatic and Invasive Plants. http://plants.ifas.ufl.edu/node/338

2.5 Egeria

Bini LM and SM Thomas. 2005. Prediction of *Egeria najas* and *Egeria densa* occurrence in a large subtropical reservoir (Itaipu Reservoir, Brazil-Paraguay). Aquatic Botany 83:227-238.

California Department of Boating and Waterways. 2000. Draft environmental impact report for the *E. densa* control program. Vol. II: Research Trial Reports.

Cook CDK and K Urmi-König. 1984. A revision of the genus *Egeria* (Hydrocharitaceae). Aquatic Botany 19:73-96.

Getsinger KD and CR Dillon. 1984. Quiescence, growth and senescence of *E. densa* in Lake Marion. Aquatic Botany 20:329-338.

Pennington TG. 2007. Seasonal changes in allocation, growth, and photosynthetic responses of the submersed macrophyte *Egeria densa* Planch. (Hydrocharitaceae) from Oregon and California. PhD dissertation. Portland, OR: Environmental Sciences and Resources, Portland State University.

Sytsma, MD and TG Pennington. 2015. Chapter 3: Vectors for spread of invasive freshwater vascular plants with a North American analysis. In Biological Invasions in Changing Ecosystems: Vectors, Ecological Impacts, Management and Predictions (Canning-Close J, ed.), 55-74. Berlin: Walter de Gruyter GmbH & Co KG.

USDA NRCS. 2009. The PLANTS Database. http://www.plants.usda.gov/

2.6 Fanwort and Cabomba

Bultemeier BW. 2009. The response of three cabomba populations to herbicides and environmental parameters: Implications for taxonomy and management. MS thesis; University of Florida.

Fasset NC. 1953. A monograph of *Cabomba*. Castanea 18(4):116-128.

Orgaard M. 1991. The genus *Cabomba* (Cabombaceae) – A taxonomic study. Nordic J Botany 11:179-203.

2.7 Starry Stonewort

Brainard AS and KI Schulz. 2017. Impacts of the cryptic macroalgal invader, *Nitellopsis obtusa*, on macrophyte communities. Freshwater Sci 36(1):55-62.

Kipp RM, M McCarthy, A Fusaro and IA Pfingsten. 2019. *Nitellopsis obtusa* (Desvaux in Loiseleur) J. Groves, (1919): U.S. Geological Survey, Nonindigenous Aquatic Species Database, Gainesville, FL. https://nas.er.usgs.gov/queries/FactSheet.aspx?SpeciesID=1688

Pullman GD and G Crawford. 2010. A decade of starry stonewort in Michigan. Lakeline: Summer 36-42.

Sleith RS, AJ Havens, RA Stewart and KG Karol. 2015. Distribution of *Nitellopsis obtusa* (Characeae) in New York, USA. Brittonia 67(2):166-172.

2.8 Parrotfeather

Invasive Plant Atlas of New England. https://www.eddmaps.org/ipane/ipanespecies/aquatics/Myriophyllum_aquaticum.htm

Mississippi State University, Geosystems Research Institute. https://www.gri.msstate.edu/ipams/FactSheets/Parrotfeather.pdf

North Carolina State University Agricultural Chemicals Manual. https://content.ces.ncsu.edu/north-carolina-agricultural-chemicals-manual/chemical-weed-control

University of Florida Center for Aquatic and Invasive Plants. https://plants.ifas.ufl.edu/plant-directory/myriophyllum-aquaticum/

United States Department of Agriculture, PLANTS Database. https://plants.usda.gov/core/profile?symbol=myaq2

United States Geological Survey, Non-indigenous Aquatic Species Database. https://nas.er.usgs.gov/queries/factsheet.aspx?SpeciesID=235

2.9 Floatinghearts

Burks KC. 2002. *Nymphoides cristata* (Roxb.) Kuntze, a recent adventive expanding as a pest plant in Florida. Castanea 67(2):206-211. http://www.jstor.org/stable/4034197

FDACS DPI. 2016. Florida Noxious Weeds List. https://www.flrules.org/gateway/RuleNo.asp?id=5B-57.007

USDA NRCS PLANTS Database. 2019 (access date). *Nymphoides* genus page. https://plants.sc.egov.usda.gov/core/profile?symbol=NYMPH2
Wunderlin RP, BF Hansen, AR Franck and FB Essig. 2019. Atlas of Florida plants. http://florida.plantatlas.usf.edu/

2.10 Waterchestnut

Crow GE and CB Hellquist. 2000. Aquatic and wetland plants of northeastern North America. University of Wisconsin Press.
Hummel M and E Kiviat. 2004. Review of world literature on water chestnut with implications for management in North America. J Aquatic Plant Manage 42:17-28. http://www.apms.org/japm/vol42/v42p17.pdf.
Invasive plants of the eastern United States website. https://wiki.bugwood.org/Archive:BCIPEUS/Trapa_natans
IPANE. 2001. Invasive Plant Atlas of New England (IPANE) at the University of Connecticut online database. http://invasives.eeb.uconn.edu/ipane/
Kiviat E. 1993. Under the spreading water-chestnut. News from Hudsonia 9(1):1-6.
Lui K, M Butler, M Allen, E Snyder, J da Silva, B Brownson and A Ecclestone. 2010. Field guide to aquatic invasive species: Identification, collection and reporting of aquatic invasive in Ontario waters. Ministry of Natural Resources, Ontario, Canada. 201 pp

2.11 Waterhyacinth

Anonymous. 2003. Flora of North America: *Eichhornia crassipes*. FNA 26:39-41. http://www.efloras.org/florataxon.aspx?flora_id=1&taxon_id=200027394
Buker GE. 1982. Engineers vs. Florida's green menace. The Florida Historical Quarterly. April 1982, pp. 413-427.
Cook CDK, BJ Gut, EM Rix, J Schneller and M Seitz. 1974. Water plants of the world: a manual for the identification of the genera of freshwater macrophytes. Dr. W Junk b.v., Publishers, The Hague.
Gopal B. 1987. Aquatic plant studies 1: water hyacinth. Elsevier, Amsterdam, The Netherlands.

2.12 Waterlettuce

Evans JM. 2013. *Pistia stratiotes* L. in the Florida peninsula: biogeographic evidence and conservation implications of native tenure for an 'invasive' aquatic plant. Conservation and Society 11:233-246. http://www.conservationandsociety.org/text.asp?2013/11/3/233/121026
USDA NRCS. 2013. The PLANTS Database: Plants profile for *Pistia stratiotes* (water lettuce). National Plant Data Team, Greensboro, NC 27401-4901 USA. http://plants.usda.gov/core/profile?symbol=PIST2&mapType=nativity

2.13 Giant and Common Salvinia

Holm LG, DL Plucknett, JV Pancho and JP Herberger. 1977. The world's worst weeds: distribution and biology. University Press of Hawaii.
Knutson, A and J Nachtrieb (eds.). 2012. A guide to mass rearing the salvinia weevil for biological control of giant salvinia. Texas A&M University AgriLife Extension Service Species Publications ESP-475. http://lubbock.tamu.edu/files/2015/06/Mass_Rearing_Salvinia_Weevil-10-15-2012.pdf
McFarland DG, LS Nelson, MJ Grodowitz, RM Smart and CS Owens. 2004. *Salvinia molesta* D.S. Mitchell (giant salvinia) in the United States: a review of species ecology and approaches to management. Aquatic Plant Control Research Program ERDC/EL SR-04-2. https://apcrp.el.erdc.dren.mil/elpubs/pdf/srel04-2.pdf
McIntosh D, C King and K Fitzsimmons. 2003. Tilapia for biological control of giant salvinia. J Aquatic Plant Manage 41:28-31. http://www.apms.org/japm/vol41/v41p28.pdf
Nelson, LS, JG Skogerboe and KD Getsinger. 2001. Herbicide evaluation against giant salvinia. J Aquatic Plant Manage 39:48-53. https://www.apms.org/wp/wp-content/uploads/2012/10/v39p48.pdf
Oliver JD. 1993. A review of the biology of giant salvinia (*Salvinia molesta* Mitchell). J Aquatic Plant Manage 31:227-231. http://www.apms.org/japm/vol31/v31p227.pdf
Sartain, BT and CR Mudge. 2018. Evaluation of 12 foliar applied non-aquatic herbicides for efficacy against giant salvinia (*Salvinia molesta*). J Aquatic Plant Manage 56:101-112.
Websites with information on giant and common salvinia: https://nas.er.usgs.gov/queries/FactSheet.aspx?speciesID=298
https://nas.er.usgs.gov/queries/FactSheet.aspx?speciesID=297
https://www.invasivespeciesinfo.gov/profile/giant-salvinia
https://plants.usda.gov/core/profile?symbol=SAMO5
https://plants.usda.gov/core/profile?symbol=SAMI7

2.14 Duckweed and Watermeal: The World's Smallest Flowering Plants

Armstrong WP. 2011. The Wayne's Word. http://waynesword.palomar.edu/1wayindx.htm

Koschnick TJ, WT Haller and L Glasgow. 2006. Documentation of *Landoltia punctata* resistance to diquat. Weed Sci 54: 615-619.

Landolt E. 1992. Lemnaceae: Duckweed family. J Arizona-Nevada Academy of Sci 26: 10-14.

Les DH, E Landolt and DJ Crawford. 1997. Systematics of the Lemnaceae (duckweeds): inferences from micromolecular and morphological data. Plant Systematics and Evolution 204:161-177.

Meijer LE and DL Sutton. 1987. Influence of plant position on growth of duckweed. J Aquatic Plant Manage 25: 28-30.

United States Department of Agriculture, Natural Resources Conservation Service. 2013. http://plants.usda.gov/

2.15 Phragmites: Common Reed

Common reed: *Phragmites australis*. University of Florida Center for Aquatic and Invasive Plants. http://plants.ifas.ufl.edu/node/323

How does it spread? Great Lakes Phragmites Collaborative. https://www.greatlakesphragmites.net/phragbasics/spread/

Phragmites australis. United States Department of Agriculture. US Forest Service. http://www.fs.fed.us/database/feis/plants/graminoid/phraus/all.html

Phragmites field guide: distinguishing native and exotic forms of common reed (*Phragmites australis*) in the United States. USDA – Natural Resources Conservation Service. https://www.nrcs.usda.gov/Internet/FSE_PLANTMATERIALS/publications/idpmctn11494.pdf

Protecting North America's wetlands from common reed. CABI. https://www.cabi.org/projects/project/56397

2.16 Purple Loosestrife

Brown CJ, B Blossey, JC Maerz and SJ Joule. 2006. Invasive plant and experimental venue affect tadpole performance. Biol Invasions 8:327-338.

Control of perennial invasive weeds requires repeated herbicide applications. University of Nebraska-Lincoln. 2018. https://cropwatch.unl.edu/2018/control-perennial-invasive-weeds-repeated-applications-herbicides

Invasive plants of the eastern United States website. https://wiki.bugwood.org/Archive:BCIPEUS/Lythrum_salicaria

Invasive species: purple loosestrife (*Lythrum salicaria*). Wisconsin Department of Natural Resources website. https://dnr.wi.gov/topic/Invasives/fact/PurpleLoosestrife.html

Muenscher WC. 1967. Aquatic plants of the United States. Cornell University Press.

Purple loosestrife (*Lythrum salicaria*) in the Chesapeake Bay watershed: a regional management plan. 2004. http://www.anstaskforce.gov/Species%20plans/doc-Purple_Loosestrifel_Mgt_Plan_5-04.pdf

Purple loosestrife: what you should know, what you can do. Minnesota Sea Grant Program (aquatic species) website. http://www.seagrant.umn.edu/ais/purpleloosestrife_info

2.17 Flowering Rush

Crow GE and CB Hellquist. 2000. Aquatic and wetland plants of northeastern North America. University of Wisconsin Press.

Harms N E, J F Shearer. Apparent herbivory and indigenous pathogens of invasive flowering rush (*Butomus umbellatus* L.) in the Pacific Northwest. ERDC/TN APCRP-BC-35. 2015

Madsen J D, B Sartain, G Turnage, M Marko. Herbicide trials for management of flowering rush in Detroit Lakes, Minnesota for 2012. Geosystems Research Institute, Mississippi State University. Report 5059. 2013.

Michigan State. Status and strategy for flowering rush (*Butomus umbellatus* L.) management. https://www.mishigan.gov/documents/deq/wrd-ais-butomus-umbellatus-499876-7.pdf

Minnesota Sea Grant Aquatic Invasive Species website. http://www.seagrant.umn.edu/ais/floweringrush

Oregon Department of Agriculture, Plant Division, Noxious Weed Control website. http://www.oregon.gov/ODA/PLANT/WEEDS/profile_floweringrush.shtml

Rice P, V Dupuis and S Ball. Flowering rush: an invasive aquatic macrophyte infesting the flathead basin (PowerPoint). http://www.weedcenter.org/Newsletter/rice_floweringrush_sshow.pdf

Turnage G, B Alcott, T Guetter. Adaptive management of flowering rush using the contact herbicide diquat in Detroit lakes, Minnesota 2016 – Final Report. Geosystems Research Institute, Mississippi State University. Report 5076. 2018.

University of Florida Center for Aquatic and Invasive Plants http://plants.ifas.ufl.edu/node/75

USDA NRCS. 2009. The PLANTS Database. National Plant Data Center, Baton Rouge, LA. http://www.plants.usda.gov/java/profile?symbol=BUUM

US Forest Service Invasive Plants website. http://www.na.fs.fed.us/fhp/invasive_plants/weeds/flowering-rush.pdf

Wersal R M, A G Poovey, J D Madsen, K D Getsinger, C R Mudge. Comparison of late-season herbicide treatments for control of emergent flowering rush in mesocosms. J. Aquatic Plant Manage 52: 85-89

2.18 Ecology and Management of Algae and Harmful Algal Blooms

Canfield DE, RW Bachmann and MV. Hoyer. 2018. Long-term chlorophyll trends in Florida lakes. J Aquatic Plant Manage 56:47-56.

Guo MX, PJ Harrison and FJR Taylor. 1996. Fish kills related to *Prymnesium parvum* N. Carter (Haptophyta) in the People's Republic of China. J Applied Phycology 8:111-117.

McGregor GB and BC Sendall. 2015. Phylogeny and toxicology of *Lyngbya wollei* (Cyanobacteria, Oscillatoriales) from north-eastern Australia, with a description of *Microseira* gen. nov. J Phycolology 51(1):109-119.

Paul VJ. 2008. Global warming and cyanobacterial harmful algal blooms. In: Cyanobacterial harmful algal blooms: State of the science and research needs, pages 239-257.

Rodgers JH, Jr., BM Johnson and WM Bishop. 2010. Comparison of three algaecides for controlling the density of *Prymnesium parvum*. J Amer Water Resources Assoc 46:153-160.

Sager D, L Fries, L Singhurst and G Southard (eds.). 2007. Guidelines for golden alga *Prymnesium parvum* management options for ponds and small reservoirs (public waters) in Texas. Texas Parks and Wildlife (Inland Fisheries): Austin, TX.

USEPA. 2011. http://cfpub.epa.gov/npdes/pesticides/pgp.cfm

3.1 A Manager's Definition of Aquatic Plant Control

(no additional resources provided)

3.2 Developing a Lake Management Plan

Cover techniques: point intercept (species composition and distribution in the whole lake) http://el.erdc.usace.army.mil/elpubs/pdf/apcmi-02.pdf

Cover techniques: line intercept (species composition and distribution in a study plot) http://el.erdc.usace.army.mil/elpubs/pdf/apcmi-02.pdf

Abundance techniques: biomass (species composition and abundance) http://www.hpc.msstate.edu/publications/docs/2007/01/3788JAPM_45_31_34_2007.pdf

Hydroacoustic techniques: SAVEWS (distribution and abundance; no discrimination among species) http://www.erdc.usace.army.mil/pls/erdcpub/docs/erdc/images/SAVEWS.pdf

Remote sensing: satellite, aircraft (distribution of plants near the surface only; no discrimination among species) http://rsl.gis.umn.edu/Documents/FS7.pdf

Rockwell HW Jr. 2003. Summary of a survey of the literature on the economic impact of aquatic weeds. Aquatic Ecosystem Restoration Foundation, Lansing, MI. http://www.aquatics.org/pubs/economic_impact.pdf

3.3 The Endangered Species Act

(additional resources are listed in the Section for clarity)

3.4 Cultural and Physical Control of Aquatic Weeds

Center for Aquatic and Invasive Plants. University of Florida IFAS website. http://plants.ifas.ufl.edu/

Krischik VA. Managing aquatic plants in Minnesota lakes. University of Minnesota Extension website. http://www.extension.umn.edu/distribution/horticulture/DG6955.html

Lembi CA. Aquatic plant management. Purdue University. http://www.ces.purdue.edu/extmedia/ws/ws_21.pdf

Maine Department of Environmental Protection Invasive Aquatic Plants website. http://www.state.me.us/dep/blwq/topic/invasives/

New Hampshire Department of Environmental Services, Volunteer Weed Watcher Program website. http://des.nh.gov/organization/divisions/water/wmb/exoticspecies/weed_watcher.htm

Vermont Department of Environmental Conservation, Water Quality Division, Aquatic Invasive Species website. http://www.vtwaterquality.org/lakes/htm/ans/lp_ans-index.htm

Wagner KJ. 2004. The practical guide to lake management in Massachusetts. http://www.mass.gov/dcr/waterSupply/lakepond/downloads/practical_guide.pdf

Washington Department of Ecology, Aquatic Plant Management website. http://www.ecy.wa.gov/programs/wq/plants/management/index.html

3.5 Mechanical Control of Aquatic Weeds

Haller WT and DK Jones. 2012. Technology and improved efficacy of mechanical control of hydrilla. Aquatics 34(3):17-19.

Haller WT, JV Shireman and DF DuRant. 1980. Fish harvest resulting from mechanical control of hydrilla. Trans Amer Fisheries Soc 109(5):517-520.

McGehee JT. 1979. Mechanical hydrilla control in Orange Lake, Florida. J Aquatic Plant Manage 17:58-61.

National Research Council. 1976. Making aquatic weeds useful: Some perspectives for developing countries. Washington, DC. 191p. https://doi.org/10.17226/19948.

http://www.ecy.wa.gov/Programs/wq/plants/management/aqua026.html

http://www.co.thurston.wa.us/stormwater/Lakes/Long%20Lake/Long_Harvesting.htm

3.6 Introduction to Biological Control of Aquatic Weeds

Biological control for the public. http://everest.ento.vt.edu/~kok/Text_frame1.htm

Biological control of weeds – it's a natural! http://www.wssa.net/Weeds/Tools/Biological/BCBrochure.pdf

Biological control of weeds: why does quarantine testing take so long? http://ipm.ifas.ufl.edu/applying/methods/biocontrol/quarantinetest.shtml

Harley KLS and IW Forno. 1992. Biological control of weeds: a handbook for practitioners and students. Inkata Press, Melbourne, Australia.

How scientists obtain approval to release organisms for classical biological control of invasive weeds. http://edis.ifas.ufl.edu/IN607

Mentz KM. 1987. The role of economics in the selection of target pests for a biological control program in the South-west Pacific, pp. 69-85. In P Ferrer and DH Stechman (eds.), Biological control in the South-west Pacific. Report on an international workshop, Vaini, Tonga, 1985. Government Printing Office, Tonga.

Plant management in Florida waters – biological control. http://plants.ifas.ufl.edu/guide/biocons.html.

Smith RF and R van den Bosch. 1967. Integrated control, pp. 295-340. In WW Kilgore and RL Doutt (eds.). Pest control – biological, physical and selected chemical methods. Academic Press, New York.

3.6.1 Insects for Biocontrol of Aquatic Weeds

Center TD, FA Dray Jr., GP Jubinsky and MJ Grodowitz. 1999 (issued 2002). Insects and other arthropods that feed on aquatic and wetland plants. U.S. Department of Agriculture, ARS Technical Bulletin No. 1870.

Forno IW and MH Julien. 2000. Success in biological control of weeds by arthropods, pp. 159-187. In: G. Gurr and S. Wratten (eds.), Biological control: Measures of success. Kluwer Academic Publishers, Dordrecht, Netherlands.

Harley KLS and IW Forno. 1992. Biological control of weeds: a handbook for practitioners and students. Inkata Press, Melbourne, Australia.

3.6.2 Grass Carp for Biocontrol of Aquatic Weeds

Canfield DE Jr. and MV Hoyer. 1992. Relations between aquatic macrophytes and the limnology and fisheries of Florida lakes. Final Report. Bureau of Aquatic Plant Management, Florida Department of Natural Resources, Tallahassee, Florida. (http://lakewatch.ifas.ufl.edu/pubs/historical/Canfield%20and%20Hoyer%201992.PDF).

Hoyer MV, MD Netherland, MS Allen and DE Canfield. 2005. Hydrilla management in Florida: A summary and discussion of issues identified by professionals with future management recommendations. Proceeding of a Special Symposium. Florida LAKEWATCH, Gainesville, FL. (https://plants.ifas.ufl.edu/wp-content/uploads/files/caip/pdfs/HydrillaMgmt_Final_June05a.pdf).

Scheffer M.1998. Ecology of shallow lakes. Chapman and Hall, London.

Shireman JV 1979; Haller W 1994. Proceedings of the grass carp symposium. Gainesville, Florida. (http://plants.ifas.ufl.edu/manage/control-methods/biological-control/chinese-grass-carp/proceedings-of-grass-carp-conferences-1979-and-1994/).

Sun J, L Wang, L Ma, F Min, T Huang, Y Zhang Z Wu and F He. 2017. Factors affecting palatability of four submersed macrophytes for grass carp *Ctenopharyngodon idella*. Environmental Science and Pollution Research 24: 28046-28054.

Zajicek PW, T Weier, S Hardin, JR Cassani and V Mudrak. 2009. A triploid grass carp risk analysis specific to Florida. J Aquatic Plant Manage 47:15-20.

3.7 Requirements for Registration of Aquatic Herbicides

(no additional resources provided)

3.7.1 Chemical Control of Aquatic Weeds

University of Florida Center for Aquatic and Invasive Plants. http://plants.ifas.ufl.edu

US Army Corps of Engineers. http://www.erdc.usace.army.mil/pls/erdcpub/docs

3.7.2 Herbicide Resistance and Resistance Management of Aquatic Plants

Benoit LK and DH Les. 2013. Rapid identification and molecular characterization of phytoene desaturase mutations in fluridone-resistant hydrilla (*Hydrilla verticillata*). Weed Sci 61:32-40.

Berger ST, MD Netherland and GE Macdonald. 2012. Evaluating fluridone sensitivity of multiple hybrid and Eurasian watermilfoil accessions under mesocosm conditions. J Aquatic Plant Manage 50:135-146.

Berger ST, MD Netherland and GE MacDonald. 2015. Development of a rapid assay to detect reduced fluridone sensitivity in invasive watermilfoils. Weed Technol 29:605– 610.

Graham RJ, JE Pratley, PD Slater and PR Bains. 1996. Herbicide resistant aquatic weeds, a problem in New South Wales. Eleventh Australian Weeds Conference Proceedings, Melbourne, Australia. 156-158.

Heap I. The International Survey of Herbicide Resistant Weeds. Available online at weedscience.org. (accessed on May 11 2020).

Hendry AP, MT Kinnison, M Heino, T Day, TB Smith, G Fitt, CT Bergstrom, J Oakeshott, PS Jørgensen, MP Zalucki, G Gilchrist, S Southerton, A Sih, S Strauss, RF Denison and SP Carroll. 2011. Evolutionary principles and their practical application. Evolution Applic 4:159-183.

Koschnick T, W Haller and L Glasgow. 2006. Documentation of landoltia (*Landoltia punctata*) resistance to diquat. Weed Sci 54:615-619.

Michel AR, RS Arias, BE Scheffler, SO Duke, M Netherland and FE Dayan. 2004. Somatic mutation-mediated evolution of herbicide resistance in the nonindigenous invasive plant hydrilla (*Hydrilla verticillata*). Molecular Ecology 13:3229–37.

Pashnick J and RA Thum. Comparison of molecular markers to distinguish genotypes of Eurasian watermilfoil, northern watermilfoil, and their hybrids. Submitted to J Aquatic Plant Manage

Taylor LA, JN McNair, P Guastello, J Pashnick and RA Thum. 2017. Heritable variation for vegetative growth rate in ten distinct genotypes of hybrid watermilfoil. J Aquatic Plant Manage 55:51-57.

Thum RA. 2018. Genetic variation and aquatic plant management: key concepts and practical implications. J Aquatic Plant Manage 56S:101-106.

Thum RA, DJ Wcisel, MP Zuellig, M Heilman, P Hausler, P Tyning, L Huberty and MD Netherland. 2012. Field documentation of decreased herbicide response by a hybrid watermilfoil population. J Aquatic Plant Manage 50:141–146.

WSSA. 1998. “Herbicide resistance” and “herbicide tolerance” defined. Weed Technol 12:789-789. doi:10.1017/S0890037X00044766

3.7.3 Spray Adjuvants: A User’s Guide

(no additional resources provided)

3.7.4 Aquatic Herbicide Application Methods

How to build weighted trailing hoses. http://plants.ifas.ufl.edu/guide/building_weighted_trailing_hoses.html

http://www.ecy.wa.gov/programs/wq/plants/management/aqua028.html

http://aquat1.ifas.ufl.edu/guide/herbcons.html

http://ohioline.osu.edu/a-fact/0015.html

http://aquatplant.tamu.edu/index.htm

University of Florida Center for Aquatic and Invasive Plants. http://plants.ifas.ufl.edu

3.8 A Discussion to Address Your Concerns: Will Herbicides Hurt Me or My Lake?

(no additional resources provided)

Glossary of terms and other information

Note: words in this glossary are defined in the context in which they are used in this manual

A

Abscission: a process in which part of a plant naturally detaches from the rest of the plant

Absorb: to soak up a substance

Acidic: having a pH of less than 7; compare to alkaline

Acre: an area containing 43,560 square feet

Acre-foot: the amount of water one foot deep in an area that covers one acre; equal to 325,851 gallons of water with a weight of approximately 2.7 million pounds; used to calculate the amount of herbicide to be applied to a body of water

Active ingredient: the specific chemical that has herbicidal activity and is responsible for killing or controlling a plant

Acute: severe or sharp, as in the shape of a leaf; or meaning rapid or quick when referring to toxicity

Adsorb: to bind to the outside or surface, such as herbicides binding to soil particles

Adsorption: the adhesion or accumulation of a substance onto another, such as herbicides binding to soil particles

Adventive: a nonnative organism that colonized an area long ago, developed a reproducing population and has become naturalized

Aeration: the introduction of oxygen to water, often accomplished with an aerator

Aerobic: containing oxygen; compare to anaerobic

Alkaline: having a pH of greater than 7; also called basic; compare to acidic

Allocation: distribution of a substance to different areas within an organism

Amphibian: an air-breathing organism that can live in terrestrial and aquatic environments

Amphipod: a small crustacean often eaten by juvenile fish

Anaerobic: lacking oxygen, as in some highly organic lake sediments; compare to aerobic

Annual: a plant that completes its entire life cycle in one year or season; compare to perennial

Anthropogenic: occurring as a result of human activity

Apical bud, apical meristem: a growing point in the uppermost portion of many plants

Arthropod: an invertebrate organism with a segmented body; examples include insects and crustaceans

Augmentation: a process where additional organisms are added to supplement existing populations; used in biocontrol

Auxin: a plant hormone that regulates growth

Axil: the area where the leaf stalk or petiole attaches to the stem

Axillary bud, lateral bud: a meristem or bud in the leaf axil or along the sides of stems; compare to apical bud

B

Ballast: weight, typically in the form of water, placed into the hull of a heavily loaded cargo ship to increase stability; usually removed or discharged when cargo is removed

Basic: see alkaline

Bathymetry: the measurement of water depths within a body of water

Benthic: relating to the bottom of a water body and the organisms that inhabit the sediments

Bioaccumulation: a process where a substance builds up in an organism after the organism consumes other organisms contaminated with the substance

Bioconcentration: the buildup of a substance in an organism at levels greater than the surrounding environment

Biocontrol: the use of an organism such as an insect or fish to control an invasive organism such as an aquatic weed

Biodiversity: a measure of the number of different species in an environment

Biomass: the amount of vegetative material (leaves, stems, etc.) produced by a plant

Biotype: an organism that differs (in appearance or another characteristic) from other organisms of the same species; sometimes referred to as a variety or subspecies

Brackish: a mixture of fresh and saline water

Bulblet: a bulb-like vegetative structure produced by some plants that is capable of forming a new plant

Bycatch: the unintentional trapping of organisms during mechanical harvesting of aquatic weeds

C

Calcified: the accumulation of calcium deposits on the leaves of a plant

Chelate: an organic compound which binds with ions such as copper

Chlorophyll: the green pigment in plants and other photosynthetic organisms that use light to produce energy

Chloroplasts: plant structures where sunlight is converted to energy

Clarity: the relative clearness of water; usually measured with a Secchi disk; compare to turbidity

Clones: organisms that are genetically identical to one another

Coevolution, coevolved: a process where different organisms in the same environment evolve or change in concert; for example, insects and plants that have evolved together over time to provide services to one another

Crown: the region of a plant where the stems and the root join together

Crustacean: an aquatic arthropod with a segmented body and hard exoskeleton; examples include lobsters, shrimp and crabs

Cuticle: a protective waxy layer that is present on the leaves of terrestrial plants but absent on the leaves of most submersed aquatic plants

Cyanobacteria: photosynthetic bacteria; also called blue-green algae

D

Deactivation: a process where a substance is rendered inactive due to a process within a plant or binding with the sediment

Defoliation: loss or removal of a plant's leaves

Degradation: breakdown of complex organic compounds into simpler substances that are then further degraded or broken down

Depuration: cleansing or purification

Desiccate: to dry out by removing most or all water from an organism

Destratification: loss of the layering that occurs in bodies of water (usually during the summer) and results in water mixing across depths within a water body; see thermocline

Detritivores: organisms that eat detritus or other dead organic matter

Detritus: decomposed organic material (primarily dead aquatic plants) that settles on and in the sediment

Dewatering: the process of removing the water from an aquatic system; see drawdown

Dicotyledon (dicot): a plant characterized by having two seed leaves at germination and leaf veins that are arranged like a net; most broad-leaved plants are dicots; compare to monocotyledon

Diluent: a substance (usually water) used to reduce the concentration of a herbicide and to facilitate uniform application

Dioecious: a condition where individual plants bear only staminate (male) or pistillate (female) flowers; compare to monoecious

Diploid: an organism with two sets of chromosomes; usually fully fertile and able to reproduce by sexual means

Dissipation: the slow reduction in concentration and eventual loss of a substance through degradation, dilution or both processes

Dormant: a condition where plants cease growth in order to survive adverse conditions and resume growth when conditions improve

Drawdown: partial or complete removal of the water in an aquatic system for a period ranging from several months to several years to cause desiccation and death of aquatic weeds

Dredge: removal of part of the sediment in a water body to improve navigation and/or control aquatic weeds; also used to describe the equipment used in this process

E

Ecosystem: the flora, fauna and environmental conditions within a given area

Efficacy: effectiveness

Embayment: a bay-shaped indentation in the shoreline that is larger than a cove but smaller than a gulf

Emergent: a plant that is rooted in the sediment with most parts of the plant maintained above the waterline; examples include most shoreline plants such as cattail, purple loosestrife and pickerelweed

Emulsifier: a substance that is used to keep particles in solution in a fluid; often added to concentrated herbicides so they can be mixed with water

Endemic: considered native or naturally occurring in an area

End-use product: the final product purchased by applicators; usually manufactured with technical grade active ingredients and diluted with inert ingredients such as water and emulsifiers to make the product easy to dilute and apply

Entomology: the study of insects

Enzyme: a chemical that degrades or breaks down a substance or allows a chemical reaction to occur

Equilibrium: a balanced system with little change in the elements that comprise the system

Eradication: complete elimination of an organism from a system; see extirpated

Estuary: the wide part of a river where it nears the ocean

Eutrophic: rich in minerals and organic nutrients; eutrophic conditions encourage algae growth and reduce levels of dissolved oxygen

Eutrophication: the accumulation of excessive minerals and organic nutrients

Evergreen: a plant that maintains its leaves and sometimes continues to grow throughout the year

Exotic: not native to a region or system

Extirpated: see eradication

F

Fauna: collectively, the animals (including insects) present in a system

Floating–leaved: a plant that is rooted in the sediment and has leaves that float on the surface of the water; examples include waterlily and waterchestnut

Flora: collectively, the plants present in a system

Formulation: the form in which a herbicide is sold (liquid, granular or other form)

Fragmentation: a process whereby part of a plant is removed from the rest of the plant due to natural (see abscission) or mechanical means

Free-floating: a plant with roots that typically occupy the upper portion of the water column; examples include waterhyacinth and salvinia

G

Generalist: an organism that does not require a specific food source for growth, survival and reproduction

Genotype: the genetic composition of an organism

Genus: a classification that describes a group of closely related organisms; each genus is further divided into species, whose members are very closely related and can breed with one another

Geotextile: a specialized fabric-like material used to stabilize shorelines or to smother submersed aquatic weeds

GLP: an acronym for good laboratory practices, a set of protocols that must be followed when testing herbicides

H

Half-life: the period of time required for the concentration of a chemical to be reduced by half, usually by microbes, light or chemical reactions

Hardness: a measure of the amount of calcium and carbonates in water

Herbaceous: a “fleshy” plant with no little or no woody material

Heterogeneity: a measure of the genetic diversity in an organism; also used to describe diverse plant communities

Heterotypic: of a different form or type

Hydrology: the study of the properties, distribution and effects of water on the earth’s surface, soil and atmosphere

Hydrolysis: the splitting of a compound into two smaller parts as a result of contact with water

Hydropower: energy derived from the force of moving water

Hypereutrophic: extremely high in nutrients; characterized by excessive algae growth that causes water to be very cloudy with poor transparency

Hypolimnetic: pertaining to the hypolimnion, the cold deeper area of a stratified lake

I

Inactivation: a process where a substance is rendered inactive due to a process within a plant or binding with the sediment

Indigenous: native to a region or system

Inert: a substance that lacks herbicidal properties

Inflorescence: the structure and arrangement of a plant’s flowers

Insectivorous: insect-eating

Inundated: flooded or under water

Invasive: a species that steals resources from desirable species and reduces diversity by being more competitive than other organisms in the system; most invasive species are nonnative, fast-growing and lack natural enemies

Invertebrate: an animal that lacks a backbone

L

LC50: abbreviation for lethal concentration 50%; the external or applied concentration of a substance required to cause death in 50% of the organisms tested; similar to LD50 (lethal dose 50%)

Larvae: early stage of insect development; examples include maggots and grubs

Lateral: a bud or branch produced from a leaf axil or other non-terminal bud on the plant

Limnology: the study of freshwater systems, including lakes, rivers and ponds

Littoral: the zone near the shoreline where water is typically shallow; usually inhabited by aquatic plants

M

Macrophyte: a plant that can be easily seen without magnification

Macroscopic: an organism that can be easily seen without magnification

Meristem: the part of a plant from which new growth originates; also called a bud

Mesotrophic: having moderate amounts of nutrients and phytoplankton

Metabolite: the product resulting from chemical breakdown or degradation of a more complex organic molecule

Microbe: a tiny organism such as a bacterium or fungus; also called microorganism

Microcrustaceans: very small zooplankton or crustaceans that feed on phytoplankton and are not easily viewed without a microscope or magnifying lens

Microfauna: animals that are not easily viewed without a microscope or magnifying lens

Micronutrient: an element that organisms require in small quantities for healthy growth

Midrib: the central vein of a leaf

Mineralization: the conversion of an element from an organic form to an inorganic form as a result of microbial decomposition

Molting: the shedding of an insect’s outer layer to allow expansion and growth

Monocotyledon (monocot): a plant characterized by having a single seed leaf at germination and leaf veins that are arranged in a parallel manner; grasses are monocots; compare to dicotyledon

Monoculture: a group of plants consisting solely of members of a single species

Monoecious: a condition where individual plants bear both staminate (male) and pistillate (female) flowers; compare with dioecious

Monotypic: composed of organisms of the same type or species

Morphology: the appearance of an organism

N

Native range: the geographic region from which an organism originates

Naturalized: a nonnative organism that reproduces and maintains a population in a new area; see adventive

Niche: a specific range of environmental conditions or a habitat in which a species can thrive

Nonindigenous: a nonnative organism

Nutlet: a small, hard, reproductive structure

O

Obligate: requiring a certain environment or food source to survive, grow and reproduce

Off-patent: a chemical that is no longer protected by a patent and can be produced by other companies in addition to the company that developed the product; often available in generic form

Oligotrophic: very low in minerals and organic nutrients

Omnivorous: consuming almost any type of plant or animal matter

Ornithology: the study of birds

Outcompete: make better or more efficient use of available resources than other organisms; deplete resources needed for growth of other organisms

Overwinter: to survive throughout the winter, often in a dormant state or as a propagule

Oxbow: a sharp, U-shaped bend in a river that is no longer attached to the river

Oxygen: present in water at concentrations ranging from 0 to 15 ppm; few fish can survive extended periods when oxygen content is below 2 ppm

Oxygenation: to increase the oxygen content of water, usually with the introduction of air into the system; see aeration

P

Palmate: arrangement where leaflets (small leaves) radiate from a central point; similar to fingers radiating from the palm of the hand

Parasite: an organism that survives by feeding on, damaging or deriving nutrients from another organism

Pathogen: an organism that causes disease to another organism

Pathology: the study of pathogens

Pelagic: referring to deep, cold water; see hypolimnion

Perennial: a plant that requires multiple years or seasons to complete its entire life cycle; compare to annual

Petiole: the "stalk" attaching a leaf to the stem of a plant

Photolysis, photolytic: the breakdown or chemical decomposition of a compound induced by light

Photosynthesis: the daytime-only process by which plants use carbon dioxide to convert sunlight into energy and oxygen

Phytoplankton: tiny, free-floating photosynthetic aquatic organisms; examples include diatoms, dinoflagellates and some species of algae

Pigment: a substance that produces a distinct color in a plant; may have protective properties

Pinnate: resembling or arranged like a feather

Piscivorous: fish-eating

Pistillate: a flower bearing female reproductive structures and lacking male reproductive structures; compare to staminate

Plankton: very small free-floating aquatic organisms; examples include phytoplankton and zooplankton

ppb: parts per billion (1 in 1,000,000,000)

ppm: parts per million (1 in 1,000,000)

Precipitate: settle as a solid to the bottom of the water body

Precipitation: a chemical reaction or process that reduces the solubility of a substance and causes it to precipitate

Predation: consumption of an organism (prey) by another organism (predator) Pristine: natural; not affected by human activity

Productivity: the trophic state of a lake (biological productivity) or the amount of organic matter produced (plant productivity)

Propagation: the act of creating new plants through sexual or vegetative means

Propagules: vegetative or sexual structures with the ability to create new plants; examples include turions, tubers, bulblets, fragments, winter buds and seeds

Protozoan: a single-celled microscopic organism; examples include amoebas and ciliates

Psyllid: an insect in the family Psyllidae; also called jumping plant lice

Pupa: the stage in insect development between larva and adult; pupae are usually protected within a hard cocoon or case

Q

Quiescence: a resting state

R

Ramet: a new plantlet formed by vegetative means; often borne on a runner or stolon

Recolonization: the re-establishment of a species that was previously found in a system but disappeared

Registrant: the organization responsible for the registration of a pesticide with the US EPA

Reservoir: a man-made body of water used for water storage, flood control, hydropower, recreation or other anthropogenic activities

Residue: any substance in food, water or an organism that occurs as a result of application of a pesticide

Resistant: the ability of an organism to survive or be unaffected by a stressor such as a herbicide; compare to susceptible

Respiration: a process in which plants take up oxygen and release carbon dioxide

Rhizome: modified plant structure that grows underground and has buds that can produce new plants

Richness: the number of distinct species present in a system

Riparian: relating to the bank or shoreline of a body of water

Rootstock: the roots, crown and rhizomes of a plant

Rosette: plant growth form where leaves radiate from a central point or crown instead of being attached to a stem

Runner: see stolon

S

Salinity: measure of the amount of salt in water

Scour: to clear a channel or remove sediment as a result of wave action, current or flow

Secchi disk: a circular disk divided into black and white sections and used to measure water clarity or transparency

Sediment: the soil or organic material at the bottom of the water body

Sedimentation: the process of accumulating sediment, usually as a result of wave action, erosion, reduced water flow in plant beds or decaying plant material

Seedbank: seeds that fall to the sediment and provide a source for new plants in future seasons

Selective: a herbicide that controls certain plants while leaving others unharmed

Senescence: plant death

Serrated: with toothed margins similar to the blade of a saw

Shoots: upright plant stems

Short-day: a condition where daylength is less than 12 hours in length (winter in the US)

Species richness: the number of different plant or animal species in a defined area

Specificity: the ability of a herbicide to selectively control target plants without causing significant damage to nontarget plants

Spores: reproductive structures produced by ferns such as salvinia

Stamen: the pollen-bearing male reproductive structure of a flower

Staminate: a flower bearing male reproductive structures and lacking female reproductive structures; compare to pistillate

Stolon: a stem-like structure or shoot that creeps along the surface of the soil or sediment; also called runner

Stratification: a layered configuration within a body of water whereby distinct and separate upper (epilimnion), middle (metalimnion) and lower (hypolimnion) layers are evident

Structure: referring to the array of architectures provided by different plants, logs, brush piles and rocks in fish habitats

Submersed: a plant that grows mostly or entirely under water

Subspecies: a division within a species to designate a group of plants that differ substantially from other members of the species

Substrate: see sediment

Surfactant: short for "surface-active agent"; a detergent-like substance that reduces surface tension and increases herbicide coverage and penetration into plant stems and leaves

Susceptible: an organism that is damaged or killed by a stressor such as a herbicide; compare to resistant

Systemic: a substance that moves throughout an organism via translocation through vessels in plants

T

Tannins: acidic yellow to brown substances derived from plant materials such as tree bark, roots, leaves and tea

Taxonomy: a system used to categorize, describe and identify organisms

Technical grade: the purest, most concentrated form of an active ingredient

Temperate: a climate that is warm in the summer and cold in the winter

Terrestrial: not flooded or inundated

Thermocline: the metalimnion or center layer of water in a stratified lake; the most extreme temperature changes occur in the thermocline as opposed to the upper (epilimnion) and lower (hypolimnion) layers

Topped-out: a phenomenon where submersed plants such as hydrilla reach the surface of the water and form dense mats or canopies that reduce penetration of light and oxygen

Toxicant: a substance used to damage or kill an organism

Translocation: active process of movement of substance within and throughout a plant

Triploid: an organism with three sets of chromosomes; usually sterile and unable to reproduce by sexual means

Trophic: related to nutrition and nutrient levels; productivity

Tuber: a vegetative propagule produced in the sediment to facilitate reproduction and overwintering

Turbidity: the degree to which water clarity is reduced by suspended particles, tannins, algae and other substances; compare to clarity

Turion: a propagule produced in the leaf axils or compressed apical buds of hydrilla to facilitate vegetative reproduction, overwintering, survival and spread

U

Upland: see terrestrial

V

Variety: a division within a species to designate a group of plants that differ substantially from other members of the species; similar to subspecies

Vascular plant: plant with a specialized internal transport or vessel system; sugars are transported in the phloem, whereas water and nutrients are transported in the xylem

Vector: an organism that transmits a disease-causing pathogen

Veliger: snail larvae

W

Watershed: the entire drainage area of a river or the catchment area of lakes

Wetland: an area that is inundated or saturated for long enough periods to support plants that are adapted to living under saturated soil conditions

Whorled: with leaves arranged in groups of three or more at a node

Winter bud: compressed apical bud; similar to turion

Z

Zonation: the separation of areas within an ecosystem into specific zones, with each zone having distinct characteristics that distinguish it from other zones

Zooplankton: microscopic aquatic animals and larvae which usually feed on phytoplankton

Common water quality parameters

Alkalinity: The water's ability to neutralize acids, measured in milligrams per liter of total alkalinity as equivalent calcium carbonate (mg/L CaCO3). Alkalinity helps regulate pH and metal content in water. Levels of 20-200 mg/L are common in fresh water systems.

Conductivity: The measure of the capacity of water to conduct an electric current, measured in either microSiemens per centimeter of water at 25 degrees centigrade (µS/cm @ 25 °C) or micromhos per centimeter (µmhos/cm). Conductivity is an indirect measure of dissolved solids such as chloride, nitrate, sulfate, phosphate, sodium, magnesium, calcium and iron.

Dissolved oxygen (DO): The amount of oxygen measured in water in milligrams per liter (mg/L). In general, rapidly moving water contains more dissolved oxygen than slow or stagnant water and colder water contains more dissolved oxygen than warmer water. Low DO levels can lead to fish kills. Optimal DO for many species is between 7 and 9 mg/L.

Hardness: Water hardness is generally the measure of the cations of magnesium and calcium in the water, usually expressed as mg/L. Waters with a total hardness in the range of 0 to 60 mg/L are termed soft; from 60 to 120 mg/L moderately hard; from 120 to 180 mg/L hard; and above 180 mg/ L very hard.

pH: Scale of values from 0 to 14 which indicate the acidity of a waterbody. Water is acidic if pH is below 7, with increasing acidity with lower values. Water is basic when above 7, and more basic with increasing values. A value of 7 is considered neutral pH. Aquatic organisms differ in the pH range they can tolerate and flourish in.

Turbidity: A measure of the amount of particulate matter that is suspended in water, and is measured in Nephelometric Turbidity Units (NTU). Water that has high turbidity appears cloudy or opaque. High turbidity can cause increased water temperatures because suspended particles absorb more heat and can also reduce the amount of light penetrating the water.

Common conversion factors

To change	To	Multiply by
acres	hectares	0.4047
acres	square feet	43,560
centimeters	inches	0.3937
centimeters	feet	0.03281
cubic feet	cubic meters	0.0283
cups	ounces (liquid)	8
cubic meters	cubic feet	35.3145
cubic meters	cubic yards	1.3079
feet/second	miles/hour	0.6818
gallons (U.S.)	liters	3.7853
grams	ounces (avdp)	0.0353
grams	pounds	0.002205
hectares	acres	2.471
inches	centimeters	2.54
kilograms	pounds (avdp)	2.2046
kilometers	miles	0.6214
liters	gallons (U.S.)	0.2642
liters	pints (liquid)	2.1134
liters	quarts (liquid)	1.0567
meters	feet	3.2808
meters	yards	1.0936
miles	kilometers	1.6093
miles	feet	5280
miles/hour	feet/minute	88
ounces (avdp)	grams	28.3495
ounces (avdp)	pounds	0.0625
ounces (liquid)	pints (liquid)	0.0625
ounces (liquid)	quarts (liquid)	0.03125
pints (liquid)	liters	0.4732
pints (liquid)	ounces (liquid)	16
pounds (avdp)	kilograms	0.4536
pounds	ounces	16
quarts (liquid)	liters	0.9463
quarts (liquid)	ounces (liquid)	32
square feet	square meters	0.0929
square kilometers	square miles	0.3861
square meters	square feet	10.7639
yards	meters	0.9144
1 ppm	= 1 mg/L or 1 mg/kg	
1 ppb	= 1 µg/L or 1 µg/kg	

More than twenty years ago, a group of companies formed a nonprofit foundation to address increasing problems with invasive aquatic weeds in complex, multiple-use ecosystems.

The mission of the AERF is to support research and development which provides strategies and techniques for the environmentally and scientifically sound management, conservation and restoration of aquatic ecosystems. Our research provides the basis for the effective control of nuisance and invasive aquatic and wetland plants and algae. Broad strategic goals include:

1. Providing information to the public on the benefits of conserving aquatic ecosystems. This involves various operationally sound methods which are appropriate for a particular water body to achieve the objectives of a sound management plan. This includes the appropriate use of EPA registered aquatic herbicides and algaecides. The foundation has produced **Biology and Control of Aquatic Plants: A Best Management Practices Manual**, which has become one of the most widely read and used references in the aquatic plant management community. This document can be downloaded from our web site (www.aquatics.org), and illustrates the various ways that aquatic plants can be managed – biological, mechanical, physical, chemical, etc.

2. Providing information and resources to assist regulatory agencies and other entities making decisions that impact aquatic plant management. This goal is partially accomplished by providing independent experts on request to address specifically defined issues. Similarly, AERF has sponsored seminars and symposia throughout the United States on aquatic plant management issues. AERF also assists state and local agencies by providing travel grants for regulatory personnel to participate in aquatic-related professional meetings.

3. Funding research in applied aquatic plant management. AERF has funded ecosystem-related research by independent scientists and graduate students in 20 universities in the United States and with the US Army Corps of Engineers. AERF also promotes the attendance of students at aquatic-related professional meetings by providing assistantships and travel grants to dozens of students annually.

Funding is generated through contributions, sponsorships, donations and grants. The operation of the Foundation is managed by an Executive Director. A Board of Directors, composed of sponsors, provides guidance on the development of annual objectives and the management of fiscal resources. Decisions are made by the Executive Director, such as the selection of subject matter experts, speakers for symposia, AERF participation in seminars and meetings and similar activities that fall within the objectives of the Foundation. A Scientific Advisory Committee composed of PhD researchers comments on the soundness of the science in the research proposals and consistency in terms of the Foundation's mission statement.

Carlton Layne, Executive Director
Aquatic Ecosystem Restoration Foundation
clayne@aquatics.org • www.aquatics.org

Restoration Regulatory Research

Your AERF sponsorship is key to

Maintaining critical efforts in education and outreach
Supporting high-quality research
Expanding partnerships with regulatory agencies
Attracting graduate students
Building partnerships
Expanding an already diverse sponsorship
Being a source for resource management

To donate to or to become a sponsor of the AERF, please send this completed application form and donation to **Treasurer, AERF, 1860 Bagwell Street, Flint, MI 48503-4406**

Please use the following as a guide in the selection of the desired level of sponsorship. Of course, you are welcome to sponsor AERF at any level and additional donations are appreciated.

Date: __________ Name: __________________________ Company: ____

Address: ____

Phone: ___________________ Fax: ___________________ Email: ____

Website: ____________________________________

☐ Check here if you would like to receive a free copy of the book with your sponsorship.

☐ Gold is recommended for manufacturers and registrants ____ $15,000

☐ Silver and above is recommended for formulators ____ $5,000

☐ Bronze and above is recommended for distributors ____ $2,500

☐ Affiliate and above is recommended for consultant and application companies, equipment manufacturers/resellers and biological producers/resellers ____ $1,000

☐ Associate and above is recommended for societies, federal and state agencies, aquatic resource management associations, applicators and consultants ____ $250

☐ Individual and above is recommended for individual sponsors _______________________________________ $50

☐ Student and above is recommended for students ____ FREE!

For more information contact:
Carlton R. Layne, Executive Director
Aquatic Ecosystem Restoration Foundation
3272 Sherman Ridge Drive, Marietta, GA 30064
Phone: 678-773-1364 • Fax 770-499-0158 • Email clayne@aquatics.org
www.aquatics.org