

Hydrilla

2014

Integrated Management

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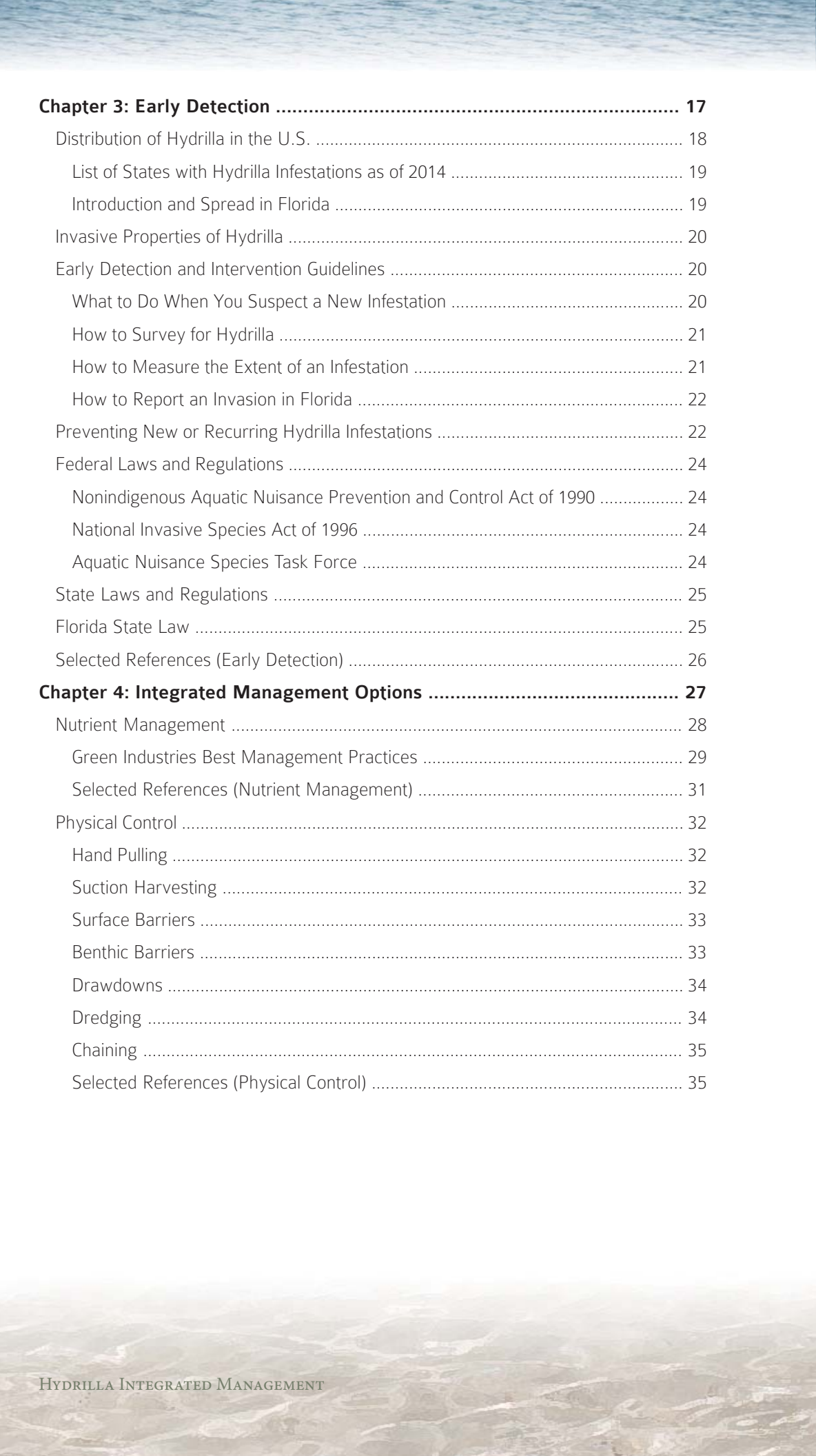
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Contents

Chapter 1: Introduction	1
How to Use this Guide	2
About Hydrilla	3
Why Can Hydrilla Be Bad for Florida’s Ecosystems?	3
Goals of Hydrilla Management	4
Integrated Pest Management (IPM)	4
Developing an IPM Plan for Aquatic Weeds	5
PAMS IPM	6
Summary Table: Methods for Hydrilla IPM	7
Selected References (IPM)	8
Chapter 2: How to Identify Hydrilla	9
Description of Hydrilla	10
Habitat	10
Plant Habit	10
Canopy	10
Stems	12
Leaves	12
Turions	12
Flowers	13
Roots	13
Tubers	13
Look-alikes	14
Canadian or Common Waterweed	14
Brazilian Waterweed	14
Summary: Hydrilla Identification	15
Selected References (Hydrilla Identification)	16



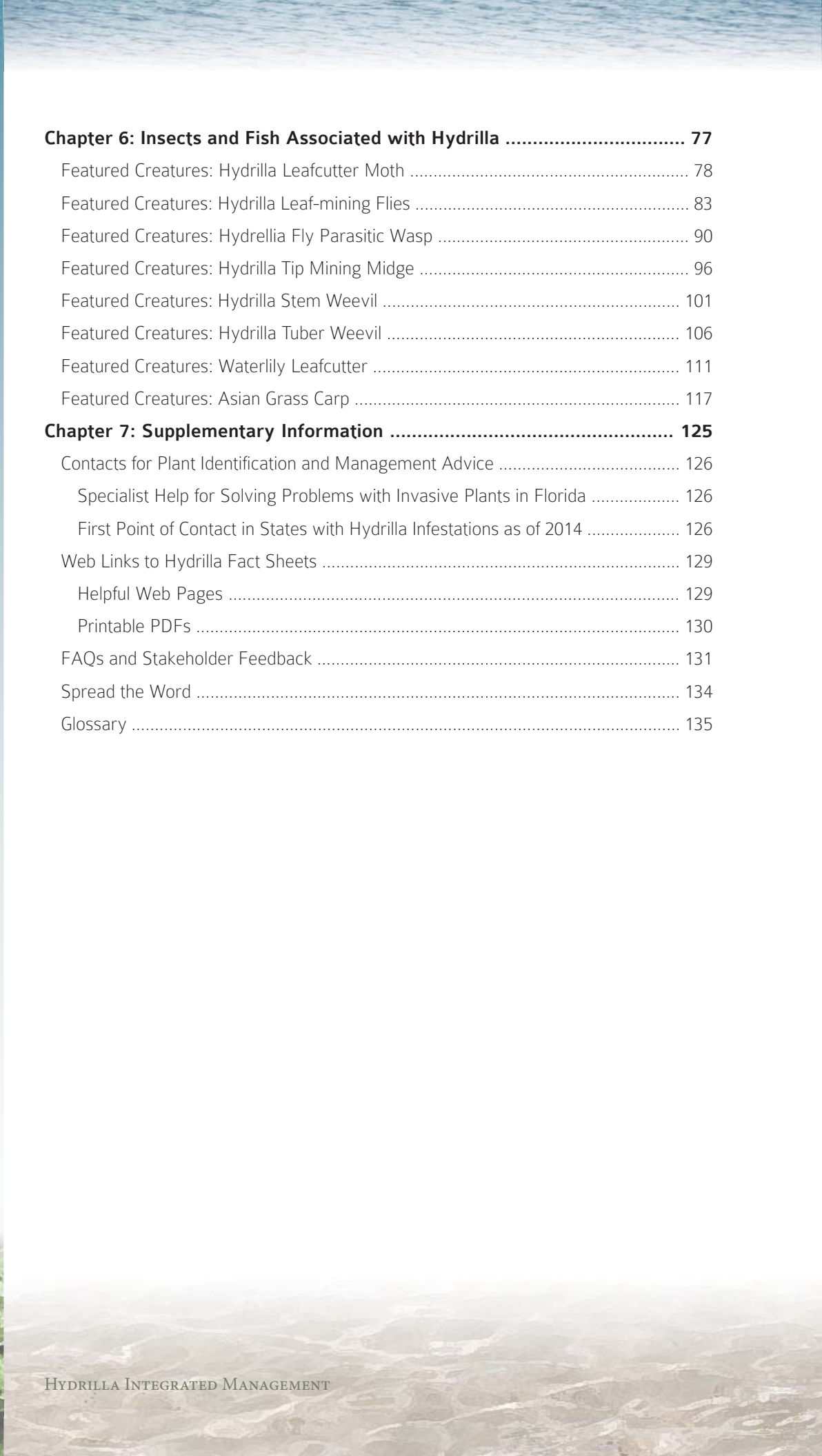
Chapter 3: Early Detection	17
Distribution of Hydrilla in the U.S.	18
List of States with Hydrilla Infestations as of 2014	19
Introduction and Spread in Florida	19
Invasive Properties of Hydrilla	20
Early Detection and Intervention Guidelines	20
What to Do When You Suspect a New Infestation	20
How to Survey for Hydrilla	21
How to Measure the Extent of an Infestation	21
How to Report an Invasion in Florida	22
Preventing New or Recurring Hydrilla Infestations	22
Federal Laws and Regulations	24
Nonindigenous Aquatic Nuisance Prevention and Control Act of 1990	24
National Invasive Species Act of 1996	24
Aquatic Nuisance Species Task Force	24
State Laws and Regulations	25
Florida State Law	25
Selected References (Early Detection)	26
Chapter 4: Integrated Management Options	27
Nutrient Management	28
Green Industries Best Management Practices	29
Selected References (Nutrient Management)	31
Physical Control	32
Hand Pulling	32
Suction Harvesting	32
Surface Barriers	33
Benthic Barriers	33
Drawdowns	34
Dredging	34
Chaining	35
Selected References (Physical Control)	35



Mechanical Control	36
Mechanical Harvesting	36
Considerations	36
Selected References (Mechanical Control)	38
Biological Control	39
Herbivorous Fish	39
Herbivorous Insects	43
Pathogenic Fungus	47
Summary Table: Potential Biological Control Agents of Hydrilla	48
Selected References (Biological Control)	49
Chemical (Herbicide) Control	50
History of Aquatic Herbicide Use	50
Herbicides Approved for Aquatic Use and Applied in Florida	51
Selected References (Chemical Control)	64
Chapter 5: Proposals for Integrated Hydrilla Management	65
Combining Different Tactics	66
Hydrilla Tip Mining Midge and a Plant Pathogenic Fungus	66
Hydrilla Tip Mining Midge and the Herbicide Imazamox	67
Plant Pathogenic Fungus and Herbicides	67
Selected References (Integrated Management of Hydrilla)	68
Resistance Management	69
What Is Resistance?	69
Herbicide Resistance in Hydrilla Populations	69
The Importance of Product Rotation	70
A Case Report on Integrated and Resistance Management of Hydrilla in Florida ...	70
How Can You Help Prevent Herbicide Resistance?	71
Selected References (Resistance and Its Management)	71
Management of Hydrilla in Different Water Body Types	72
Characteristics of Different Water Bodies	72
Management of Submersed Weeds in Different Waters	73
Selected References (Different Water Bodies and Weed Control)	76



Chapter 6: Insects and Fish Associated with Hydrilla	77
Featured Creatures: Hydrilla Leafcutter Moth	78
Featured Creatures: Hydrilla Leaf-mining Flies	83
Featured Creatures: Hydrellia Fly Parasitic Wasp	90
Featured Creatures: Hydrilla Tip Mining Midge	96
Featured Creatures: Hydrilla Stem Weevil	101
Featured Creatures: Hydrilla Tuber Weevil	106
Featured Creatures: Waterlily Leafcutter	111
Featured Creatures: Asian Grass Carp	117
Chapter 7: Supplementary Information	125
Contacts for Plant Identification and Management Advice	126
Specialist Help for Solving Problems with Invasive Plants in Florida	126
First Point of Contact in States with Hydrilla Infestations as of 2014	126
Web Links to Hydrilla Fact Sheets	129
Helpful Web Pages	129
Printable PDFs	130
FAQs and Stakeholder Feedback	131
Spread the Word	134
Glossary	135



Chapter 1



Introduction

By Verena-U. Lietze and
Emma N.I. Weeks

CONTENTS

How to Use this Guide	2
About Hydrilla	3
Integrated Pest Management (IPM)	4
Developing an IPM Plan	5
PAMS IPM	6
Summary Table	7
Selected References	8

Habitat — an area or environment where a plant, fungus, or animal normally lives and grows

Herbicide — a substance that kills weeds (usually a chemical compound)

Resistance [n.], resistant [adj.] (to herbicides) — the ability of a plant to survive the exposure to a typically lethal dose of herbicide

Submersed — a plant with most leaves growing underwater; flowers and some of the leaves may float on the water surface



Figure 1. Surface mats of hydrilla along the shoreline at Wacissa River Springs, Florida. The close-up in the bottom photo shows the hydrilla mats in the top photo. Photographs by Verena Lietze, University of Florida.

Throughout Florida and at least twenty-seven other states, the invasive freshwater plant hydrilla (*Hydrilla verticillata*) causes damaging infestations that choke out native plants, clog flood control structures, and impede waterway navigation and recreational use.

This guide will help you identify hydrilla, describe commonly used integrated pest management (IPM) strategies, and provide you with contact information for further assistance when you or your clientele encounter hydrilla.

“IPM is the coordinated use of pest and environment information and available pest control methods to prevent unacceptable levels of damage by the most economical means with the least possible hazard to people, property and the environment.”

— U.S. Environmental Protection Agency (EPA)

Share this guide with others!
Help us spread the word that new strategies for hydrilla integrated pest management (IPM) are being investigated and developed.

How to Use this Guide

Use this guide to identify hydrilla (Figure 1), learn about commonly used integrated pest management (IPM) strategies, and find contact information for further assistance when you or your clientele encounter hydrilla.

Be aware that any approach that manipulates the waterways in your state requires compliance with federal and state laws. It makes sense to most people that the introduction of plants is prohibited. However, please realize that the removal of plants (even if well intended) is generally not allowed without prior permission. Always contact your state's Fish and Wildlife Conservation Commission to receive appropriate guidance.

The methods described in this guide serve as a reference of options that are currently available to prevent and manage hydrilla infestations. But keep in mind that each water body is unique and needs to be assessed in order to install the best-suitable IPM plan.

Management is expensive and labor intensive, so good planning can save money and time. Consult with experts, develop an IPM plan that fits the environment and your budget, and be open to adjustments. Such adaptive management includes learning from past mistakes, considering all resources, evaluating results, and gaining new knowledge as well as achieving desirable short-term and long-term outcomes.

One highlight of this guide is a case report that illustrates how important it is to adjust and expand management plans when you are faced with changing situations in infested water bodies (see page 70). Use the report as an orientation. It always helps to learn what people have done to make management programs successful.

About Hydrilla

Every year, the state of Florida spends millions of dollars managing this weed in our waterways. However, aquatic resource managers are facing a problem: hydrilla is showing resistance to the widely used herbicides fluridone and endothall.

To tackle this problem, UF/IFAS County Extension Faculty and Entomology and Nematology Department Faculty are implementing a U.S. Department of Agriculture (USDA) grant-funded program: the Hydrilla Integrated Pest Management Risk Avoidance and Mitigation Project (Hydrilla IPM RAMP). Within the framework of this project, new approaches for managing hydrilla are being evaluated (Figure 2).

Why Can Hydrilla Be Bad for Florida's Ecosystems?

Remember that hydrilla is “good” in its native range. It is only when plants are introduced into a new habitat that they can begin disrupting the native environment. It is important to understand the negative impacts that hydrilla can have on an ecosystem before you consider techniques to include in your integrated management program.

Hydrilla, particularly when very dense and expansive, can reduce the number of other species, both flora and fauna, that are able to survive in the water body by:

- Reducing the level of dissolved oxygen available to other organisms
- Preventing sunlight penetration to other submersed plants
- Accessing nitrogen and phosphorus and limiting nutrients for use by other organisms

Hydrilla also can prevent the function of the water body by:

- Limiting water flow and causing flooding
- Preventing pumping (e.g., for irrigation or livestock watering)
- Preventing recreation, boating, swimming, fishing (Figure 3)
- Changing the relative densities of fish species available for fishing

Long term negative effects on the ecosystem include:

- Increased amount of sediment accumulation due to hydrilla decay
- Increased sedimentation over time, which—if not corrected—will cause water bodies to become shallow and eventually turn to wetlands

Finally, when hydrilla forms dense canopies at the surface, these areas provide excellent microhabitats. These habitats are known to provide mosquitoes with suitable breeding sites where the water is still and warm. Recent studies have discovered that hydrilla is a substrate for a newly discovered species of cyanobacterium, which produces a toxin that causes neurological disease in waterbirds including bald eagles and American coots.

Hydrilla is not the only plant we need to worry about. You should always be careful not to transport plants or animals to new ecosystems.

Figure 3. When hydrilla tops out, it forms a dense canopy (top) that prevents sunlight and nutrients reaching other organisms and interferes with recreational activities, such as boating (bottom), fishing and swimming. Photographs by Verena Lietze (top), University of Florida, and Don Schmitz (bottom), UF/IFAS Center for Aquatic and Invasive Plants.



Figure 2. UF/IFAS field research plot in Florida for testing biological control agents against hydrilla. The photo on the bottom shows a researcher (Dr. James Cuda) releasing larvae of the hydrilla tip mining midge into a marked transect of the hydrilla-infested water body. Photographs by Dana Denson, Reedy Creek Improvement District.

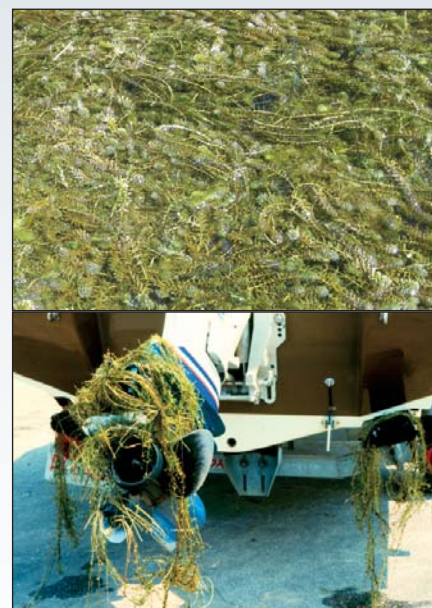


Figure 3 (see left).

Biotype — a form of the same plant species that shows special characters (for example, presence/absence of male or female flowers, resistance to a chemical herbicide, tolerance to extreme temperatures)

Habitat — an area or environment where a plant, fungus, or animal normally lives and grows

Herbicide — a substance that kills weeds (usually a chemical compound)

Resistance [n.], resistant [adj.] (to herbicides) — the ability of a plant to survive the exposure to a typically lethal dose of herbicide

Goals of Hydrilla Management

The two most important goals of every successful hydrilla management plan should be:

- To provide a sustainable, long-term, reduced-risk solution for hydrilla control
- To encourage resource managers to adopt new management tactics

What does this statement entail? Mostly: education, education, and education. Researchers all over the country are doing fantastic work to provide applicators, managers, and legislators with new data, updated methods, novel technologies, and innovative integrative approaches to alleviate the ever-increasing problems associated with invasive aquatic weeds.

Hydrilla not only has an inherently high competitive advantage over native aquatic plant species but also exhibits adaptive strategies to survive control methods. For example, the continuous application of a single effective chemical herbicide has led to the spread of herbicide-resistant biotypes of hydrilla.

In this guide, we therefore emphasize the importance of product rotation and integration of different control tactics for successful long-term management of this plant.

Integrated Pest Management (IPM)

The following definition of the term Integrated Pest Management (IPM) is given by the U.S. Environmental Protection Agency (EPA):

“IPM is the coordinated use of pest and environment information and available pest control methods to prevent unacceptable levels of damage by the most economical means with the least possible hazard to people, property and the environment.”

Coordinating the control efforts among stakeholders allows not only combining expertise but also sharing resources and tools. Stakeholders often include private, federal, state, county, and tribal entities.

Remember that IPM is based on a continuum. You can have anything from all natural and cultural controls (like native insect herbivores and hand harvesting weeds) to chemical control options. When we consider chemical control options, we often think of them as a last resort.

What distinguishes IPM from conventional pest control is that IPM goes beyond the action of going out and facing an existing problem. The principles of IPM encourage us to go one step further or, actually, take a step back and start before a problem has built up. IPM therefore includes four major steps:

1. PREVENTION: One of the most important tactics in IPM is to manage an area in a way that pests and weeds will not have the chance to become a threat. In an aquatic system, this may involve regular surveys to help detect infestations at an early stage, when manual removal is still possible and effective. Implementing control at an early stage increases the chance of eradication and decreases the costs and environmental impacts of management.

2. MONITORING AND IDENTIFYING PESTS OR WEEDS: Insects and weeds are not automatically harmful. Depending on the habitat in which they occur, they may remain at low densities and under certain circumstances even be beneficial. It is therefore important to monitor and correctly identify all species in a habitat.

3. SETTING ACTION THRESHOLDS: Some pests or weeds may not harm the environment when their density remains below a certain threshold. Action is required when this threshold is passed so that the pest or weed population will not grow out of control and cause environmental and economic damage. For invasive aquatic weed species, such as hydrilla, this threshold is very low, because these species usually have a high competitive advantage over native species and will literally take over the environment.

4. CONTROL: When steps 2 and 3 indicate that the pest or weed has become a problem that can no longer be alleviated by preventive methods, it is time to identify the best-possible control method or methods. The best-possible methods will pose the least risk to the environment and health of other organisms and provide the most effective results in an acceptable timeframe and at reasonable cost.

Developing an IPM Plan for Aquatic Weeds

The following information is based on the publication *How to Create a Lake Management Plan* by Jess M. VanDyke from a cooperative project between Florida Department of Environmental Protection and Florida LAKEWATCH.

The development of a management plan is a stepwise process:

1. Notice the infestation; identify the weed.
2. Act fast! If you only see a few weeds, perhaps suction harvesting or careful hand pulling should be done first to keep the weed from spreading out of control as you develop an action plan.
3. Form a working group of representative stakeholders from all user groups.
4. Request from all group members a list of problems associated with the infestation.
5. Arrange a meeting to assess the list and write a concise problem statement.
6. Collect information on the weed and the specific environment (water body).
7. Write a description of the water body including its environment and use.
8. List possible solutions (this is a brainstorming effort, and all ideas are allowed); be sure to list solutions that have different modes of action.
9. Evaluate each listed solution based on its expected effectiveness, longevity, confidence, applicability, potential negative impacts, capital costs, and operation/maintenance costs.
10. Based on the evaluation, refine the list of solutions.
11. Write a first draft of a management plan; be sure to include as many IPM options as possible.
12. Seek feedback from as many user groups as possible.
13. Based on the feedback, revise the management plan.
14. Find funding.
15. Implement the plan.
16. Monitor and document the results.

Documentation of the results cannot be over-emphasized. Data from monitoring will indicate when adjustments to the IPM plan may become necessary.

PAMS IPM

Do you know about PAMS? The letters stand for Prevention, Avoidance, Monitoring, and Suppression. Our information on hydrilla IPM will help you fit your management plan into the PAMS IPM model in the following ways:

PREVENTION AND AVOIDANCE—providing educational material that will help stakeholders to prevent the spread of resistant hydrilla.

MONITORING—looking for new areas where susceptible and resistant hydrilla biotypes may be introduced.

SUPPRESSION—integrating herbivory by the hydrilla tip mining midge with a fungal plant pathogen and/or low doses of a new acetolactate synthase (ALS) inhibiting herbicide as a viable strategy for long-term sustainable management of hydrilla (Figure 4).

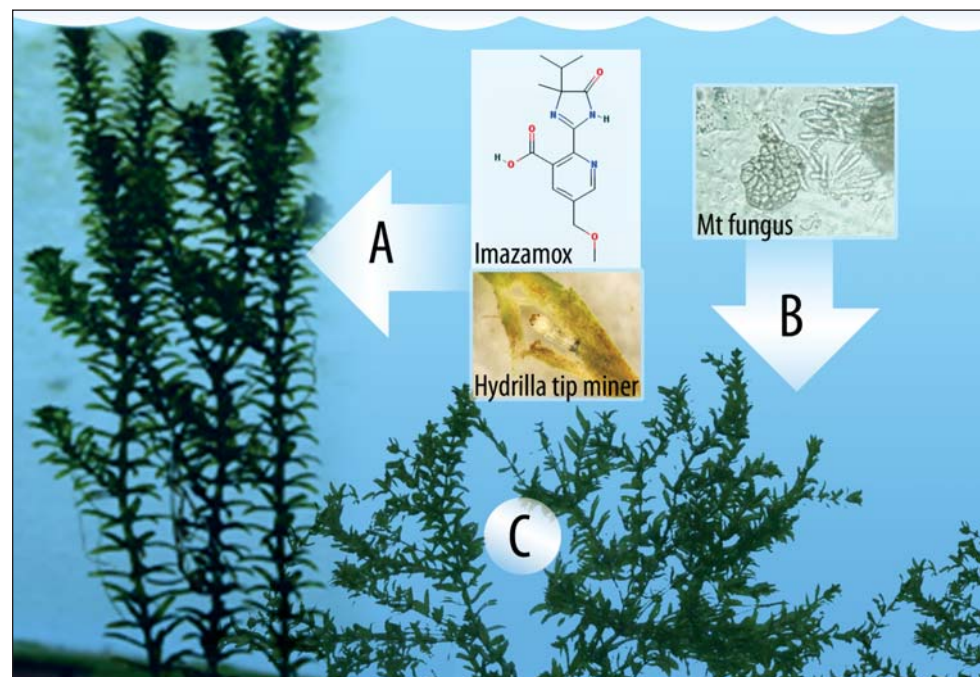


Figure 4. Expected interactions between control tactics:

A. Treating hydrilla with low concentrations of imazamox induces branching, which increases the number of breeding sites for the developing larvae of the hydrilla tip mining midge that mine hydrilla's shoot tips.

B. The mining damage changes the plant's architecture by severely injuring or killing its growing tips and increases the susceptibility of hydrilla to infection by the fungus *Mycoleptodiscus terrestris* (*Mt fungus*).

C. Combining these three tactics—an herbicide, insect, and pathogen—should prevent new hydrilla stems from reaching the surface of the water column, or topping out. Preventing hydrilla from topping out is important, so the plants cannot have a negative impact on the environment.

Biotype — a form of the same plant species that shows special characters (for example, presence/absence of male or female flowers, resistance to a chemical herbicide, tolerance to extreme temperatures)

Herbicide — a substance that kills weeds (usually a chemical compound)

Herbivorous [adj.], herbivory [n.] — plant-eating

Indigenous — occurring naturally in a place or region

Resistance [n.], resistant [adj. (to herbicides)] — the ability of a plant to survive the exposure to a typically lethal dose of herbicide

Summary Table: Methods for Hydrilla IPM

Type of management	Method	Tubers killed	Hydrilla fragments produced	Selective	Removes hydrilla	Labor intensive	Estimated cost	Pages
Nutrient	Nutrient manipulation	No	No	No	No	Yes	Depends on problem	28
Physical	Hand pulling	Possibly	No	Yes	Yes	Yes	\$400-\$1,000 per acre	32
	Suction harvesting ¹	Possibly	Possibly	Yes	Yes	Yes	\$1,000-\$25,000 per acre	32
	Surface barriers	No	No	No	No	No	\$3,200 per 100 ft length	33
	Benthic barriers ²	No	No	No	Yes	Yes	\$10,000-\$20,000 per acre	33
	Drawdowns ³	No	No	No	Yes	No	Inexpensive	34
	Dredging ¹	Yes	Yes	No	Yes	No	\$290 per mile	34
	Chaining	No	Yes	No	Yes	No	\$60 per mile	35
Mechanical	Harvesting ¹	No	Yes	No	Yes	No	\$1,000 per acre	36
	Draglines/Trackhoes ¹	No	Yes	No	Yes	No	\$290 per mile	36
Biological	Asian grass carp	No	Yes	No	Yes	No	\$15-\$150 per acre	39, 117
	Herbivorous insects	Possibly	Yes	Yes	Possibly	No	Inexpensive	43, 78
	Pathogenic fungus	No	No	Yes	Yes	No	Unknown	47
Chemical	Contact herbicides	No	No	Possibly	Yes	No	\$100-\$500 per acre	51, 54, 55
	Systemic herbicides	Possibly	No	Possibly	Yes	No	\$100-\$500 per acre	51, 54, 56

¹ Specialized equipment needed

² Usually not permitted in Florida

³ Need controlled water level

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Chapter 2

How to Identify Hydrilla

By Verena-U. Lietze and
Emma N.I. Weeks

CONTENTS

Description of Hydrilla	10
Habitat	10
Plant Habit	10
Canopy	10
Stems	12
Leaves	12
Turions	12
Flowers	13
Roots	13
Tubers	13
Look-alikes	14
Summary Diagram	15
Selected References	16

Habitat — an area or environment where a plant, fungus, or animal normally lives and grows

Submersed — a plant with most leaves growing underwater; flowers and some of the leaves may float on the water surface

This chapter will describe hydrilla and two aquatic plant species that look very similar to hydrilla.

Description of Hydrilla

The following sections will give you detailed descriptions of the hydrilla plant including its canopy on the water surface and its individual parts.

Habitat

Hydrilla grows in literally all freshwater habitats including springs, streams, rivers, lakes, ponds, reservoirs, and canals. It tolerates low light intensity, high turbidity, and a range of water qualities.

The optimum temperature for hydrilla growth is 68-81°F (20-27°C). However, hydrilla can survive at temperatures of 86°F (30°C) and is relatively cold hardy being present as far north as Maine and Washington. In many areas of the U.S., the stems and leaves die back during the winter but are quickly replaced by new growth in the spring. The turions and tubers are protected more from the cold than the stems and leaves and will survive in the sediment to produce new plants when the warm weather returns in the spring.

Hydrilla is able to grow at a lower light intensity than most other submersed aquatic weeds; it requires just 1% of full sunlight. This means that it can grow at greater depths than most other submersed plants. Hydrilla is frequently found growing in lakes at depths of 9 feet (3 m) but also is found growing 45 feet (15 m) deep in Kings Bay, Crystal River, Florida.

Nutrient conditions do not seem to impact the ability of hydrilla to infest a water body. However, the hydrilla in a water body with low nutrient levels is less likely to become dense and topped out. Hydrilla has been found growing in nutrient-rich (i.e., eutrophic) and nutrient-poor (i.e., oligotrophic) lakes. Hydrilla even tolerates salinity levels of up to 7% (equivalent to 70 parts per thousand [ppt] or 70 g/kg), which is twice as high as the average salinity level of ocean water. For comparison, the salinity of freshwater usually is less than 0.05% (0.5 ppt). Despite the ability to tolerate high salinity, hydrilla prefers freshwater environments and usually is outcompeted by other plant species in brackish water of high salinity.

Plant Habit

Hydrilla is a submerged, perennial, rooted plant with slender, vertically growing stems (Figure 5). Stems are sparsely branched until they reach the water surface, where profuse branching leads to a dense canopy.

Canopy

When the vertically growing stems reach the top 2 feet below the water surface, they begin to branch and form a dense mat of plant material. About 80% of hydrilla's biomass is in this top canopy. At this stage, hydrilla is described as being "topped out." The dense surface canopy may remain rooted or may break loose resulting in floating mats of vegetation (Figure 6).



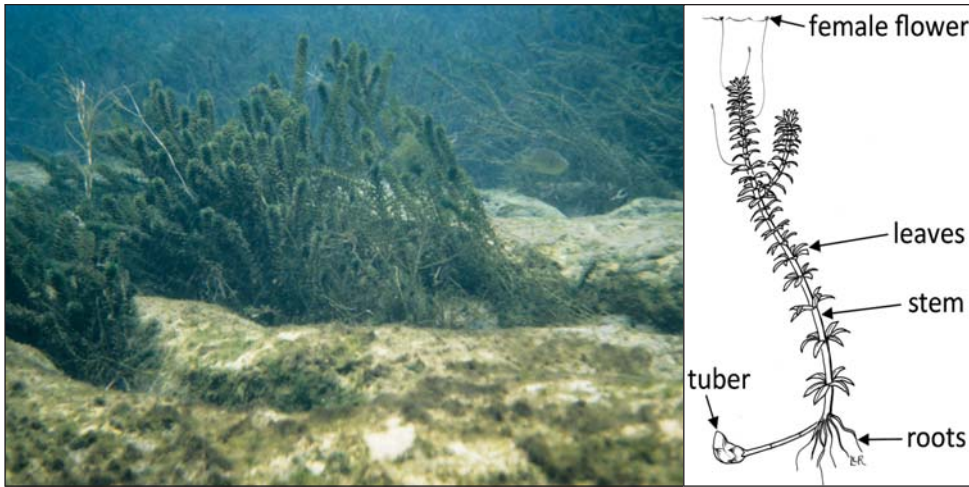


Figure 5. Hydrilla plants rooted in the sediment (left) and detailed in a line drawing (right). Photograph by Vic Ramey, UF/IFAS Center for Aquatic and Invasive Plants. Drawing by UF/IFAS Center for Aquatic and Invasive Plants.



Figure 6. Topped-out hydrilla covering the water surface of a lake in Florida. Photograph by William Haller, UF/IFAS Center for Aquatic and Invasive Plants.



Whorl (leaf whorl) — an arrangement of three or more leaves emerging and radiating from a common node along the stem

Stems

Stems (Figure 7A+B) can be up to 33 feet (10 m) long, depending on the depth of the water body. Under water, they are erect and generally not branched; at the water surface, they branch heavily. Near the sediment, stems can form stolons, which are stems that grow along or just below the surface of the sediment and produce new roots at the nodes as well as new plants from buds.

Leaves

Leaves (Figure 7C+D) are small and narrow, just about 1/10 inch (2.5 mm) wide and 1/4 to 3/4 inch (6 to 18 mm) long. They grow in whorls of 4-8 (often 5; Figure 7C) around the stem. The leaf margins are saw-toothed, and the leaf midrib carries one or more small, sharp teeth on the underside (Figure 7D).



Figure 8. Turions of hydrilla. Photographs by Lyle Buss, University of Florida.

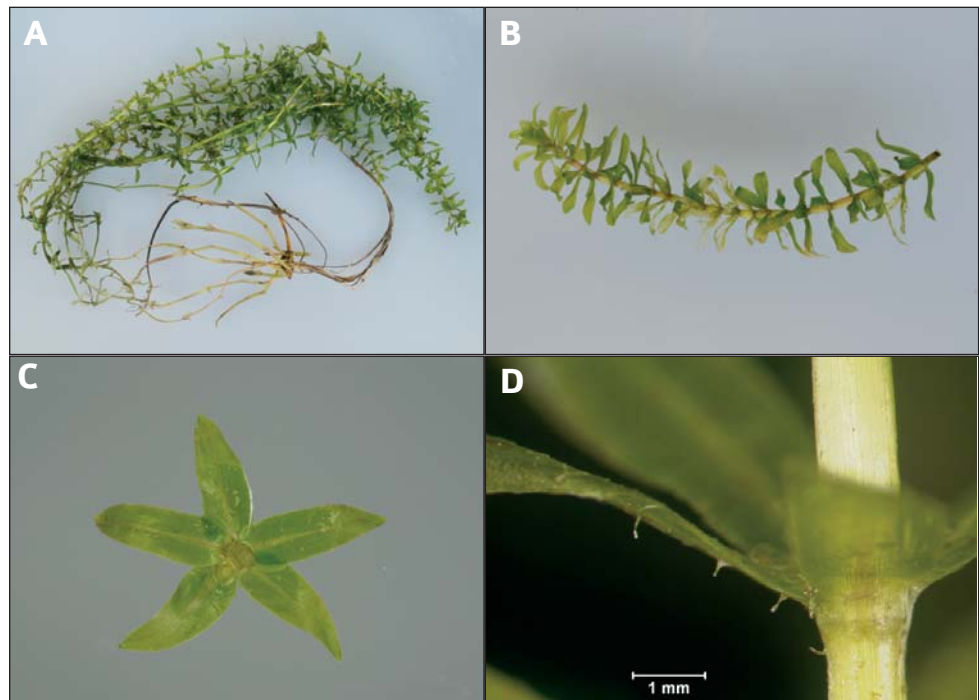


Figure 7. Stems and leaves of hydrilla. A. Lightly branched stem (green part on top) of hydrilla including the roots (brown part on bottom). B. Close-up of a growing hydrilla stem tip. C. Leaf whorl of hydrilla. D. Underside of a leaf showing the characteristic teeth. Photographs by Lyle Buss, University of Florida.

Turions

Turions (Figure 8) are buds in leaf axils. They are cylindrical in shape, about 1/4 inch (6 mm) in diameter, and dark green in color. Turions break off and fall into the sediment, where they overwinter and produce new plants in the spring.

Flowers

The female flowers (Figure 9) grow on individual long stalks to float on the water surface. Their three sepals and three petals are whitish and about 1/6 inch (4 mm) long. In Florida and the southern U.S. range of hydrilla distribution, no male flowers are found because only the female dioecious form of hydrilla is present. In northern states, the monoecious form of hydrilla (with male and female flowers on the same plant) is present.

Male flowers (Figure 9) are tiny and greenish in color, grow close to leaf axils on the shoot tips, and eventually break loose and float to the water surface.



Figure 9. Female (left photo and left drawing) and male (right photo and right drawing) flowers of hydrilla. Photographs by Matthew Purcell, U.S. Department of Agriculture, Agricultural Research Service, Australian Biological Control Laboratory CSIRO Ecosystem Sciences (left), and Michael J. Grodowitz, U.S. Army Engineer Research and Development Center (right). Drawings by UF/IFAS Center for Aquatic and Invasive Plants.

Roots

Roots (Figure 10, top) are thin and long, whitish to light brown in color, and anchored in the sediment. Roots can also form at the nodes along the stem or at the end of loose stem fragments.

Tubers

Tubers (Figure 10) are similar to turions, except that they form from rhizomes (modified below-ground stems) in the sediment. They are enlarged potato-shaped rhizomes, about 1/2 inch (12 mm) long, and yellowish brown in color. Tubers can stay viable in the sediment for several years before they sprout new shoots. They can stay viable for several days out of water. Sprouting sometimes is induced after a drawdown or a chemical treatment.

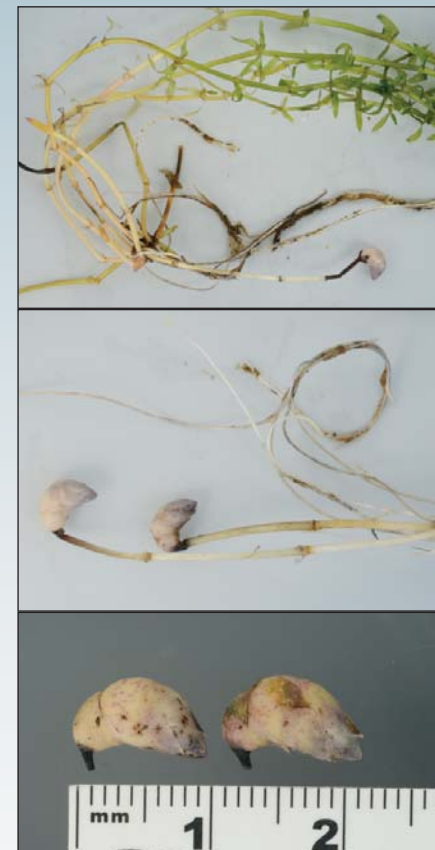


Figure 10. Roots and tubers of hydrilla. Photographs by Lyle Buss, University of Florida.

Dioecious — female and male flowers occur on different plants

Distribution — the geographical range in which the plant occurs

Monoecious — female and male flowers occur on the same plant

Look-alikes

Two aquatic plants commonly found in U.S. freshwater bodies look very similar to hydrilla. They are Canadian or common waterweed and Brazilian waterweed.

Canadian or Common Waterweed

Canadian or common waterweed (*Elodea canadensis*, Figure 11) is native to the United States. It does not produce subterranean tubers and feels softer than hydrilla because the leaves do not have teeth on the midrib. Canadian waterweed is densely whorled with two to three leaves per whorl and is considered to be beneficial at a low density.



Figure 11. Canadian or common waterweed (*Elodea canadensis*). Photographs by William Haller (left photo) and Vic Ramey (right photo), UF/IFAS Center for Aquatic and Invasive Plants.

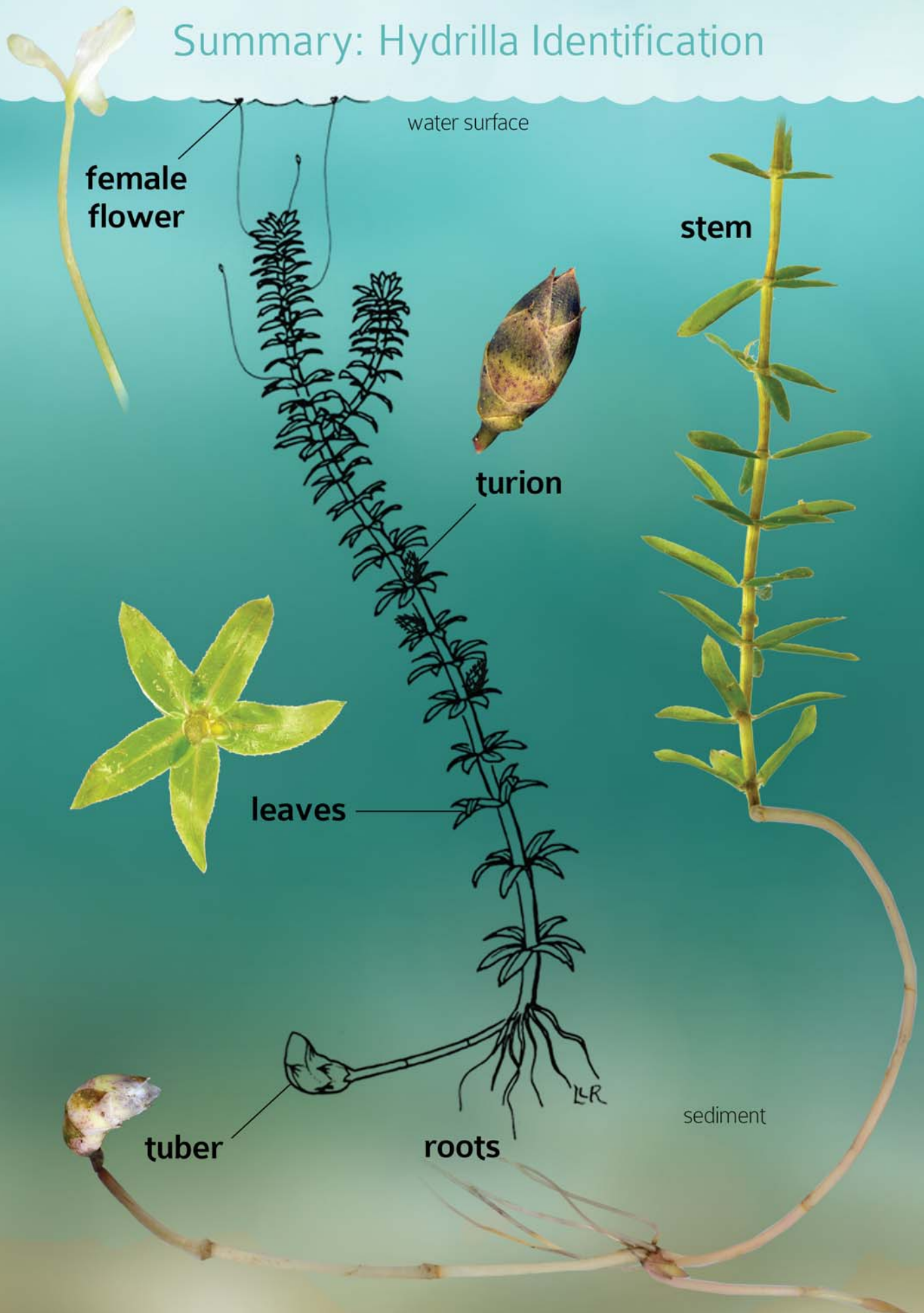
Brazilian Waterweed

Brazilian waterweed (*Egeria densa*, Figure 12) is non-native and invasive. Its leaves are about an inch (25 mm) long, finely toothed on the margins, and arranged in whorls of four to five. They carry no teeth on the midrib and feel softer than hydrilla. Brazilian waterweed does not produce tubers.



Figure 12. Brazilian waterweed (*Egeria densa*). Photographs by William Haller (left photo) and Vic Ramey (right photo), UF/IFAS Center for Aquatic and Invasive Plants.

Summary: Hydrilla Identification



Selected References (Hydrilla Identification)

Barko JW, Smart RM, McFarland DG, Chen RL. 1988. Interrelationships between the growth of *Hydrilla verticillata* (L.f.) Royle and sediment nutrient availability. *Aquatic Botany* 32: 205-216.

Glomski LM, Netherland MD. 2012. Does hydrilla grow an inch per day? Measuring short-term changes in shoot length to describe invasive potential. *Journal of Aquatic Plant Management* 50: 54-57.

Hall DW, Vandiver VV, Gray CJ. 1991. *Hydrilla, Hydrilla verticillata* (L.f.). Agronomy Department, Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida, document SP37. URL: <http://edis.ifas.ufl.edu/pdffiles/FW/FW01800.pdf> (22 April 2014).

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Haller WT, Miller JL, Garrard LA. 1976. Seasonal production and germination of hydrilla vegetative propagules. *Journal of Aquatic Plant Management* 14: 26-29.

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Chapter 3



Early Detection

By Verena-U. Lietze and
Emma N.I. Weeks

CONTENTS

Distribution of Hydrilla in the United States	18
Invasive Properties of Hydrilla	20
Early Detection and Intervention Guidelines	20
Preventing Hydrilla Infestations	22
Federal Laws and Regulations	24
State Laws and Regulations	25
Florida State Law	25
Selected References	26

Distribution — the geographical range in which the plant occurs

This chapter will describe the distribution of hydrilla in the U.S., explain why this aquatic plant has become invasive, and guide you through methods of early detection and intervention. Invasive plants are regulated on the federal and state levels, so we have summarized the most important laws for you.

Distribution of Hydrilla in the U.S.

To date (2014), at least twenty-seven states have recorded hydrilla infestations (see Figure 13), and three states are on the lookout with early eradication programs planned.

To find the first point of contact in states with hydrilla infestations, please refer to the list provided on page 126.

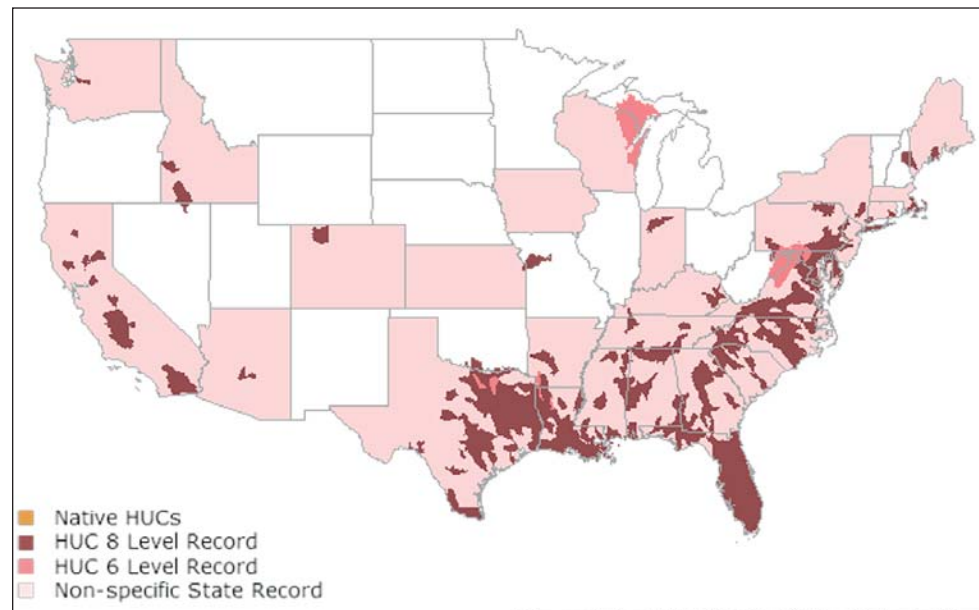


Figure 13. Distribution of hydrilla in the United States as of 2011. Distributions classified by drainage systems at two scales, Hydrologic Unit Code (HUC) 6 (= medium scale) and HUC 8 (= fine scale). HUC 6 is known as a basin and is on average 10,600 square miles in area. HUC 8 is known as a sub-basin and is on average 700 square miles in area. Occurrence of hydrilla within a drainage system results in highlighting the entire drainage. Map created by the U.S. Geological Survey, Department of the Interior/USGS, available in the public domain.

List of States with Hydrilla Infestations as of 2014

Female dioecious form	Monoecious form	Both forms	States on the lookout
Alabama (AL)	Delaware (DE)	California (CA)	Illinois (IL)
Arizona (AZ) ¹	District of Columbia (DC)	Connecticut (CT)	Michigan (MI)
Arkansas (AR)	Indiana (IN)	Georgia (GA)	Ohio (OH)
Florida (FL)	Maine (ME)	North Carolina (NC)	
Idaho (ID)	Maryland (MD)	South Carolina (SC)	
Louisiana (LA)	Massachusetts (MA)	Virginia (VA)	
Mississippi (MS)	Missouri (MO)	Wisconsin (WI)	
Tennessee (TN)	New Jersey (NJ)		
Texas (TX)	New York State (NY)		
	Pennsylvania (PA)		
	Washington (WA) ²		

¹ Eradicated from two ponds in the mid-1980s, no new infestations

² Possibly eradicated

Introduction and Spread in Florida

The female dioecious biotype of hydrilla made its way from Asia to Florida in the 1950s as an exotic aquarium plant. It was released by an aquarium owner into a freshwater body in the Tampa Bay area.

Within forty years of this introduction, hydrilla populations have established in over 70% of Florida's watersheds (Figure 14). From Florida, dioecious hydrilla has spread to most of the southeastern coastal states.

The monoecious biotype of hydrilla was first detected in Maryland in the 1970s and is thought to originate from Korea. It now is present along the eastern and western coasts as far north as Maine and Washington State, respectively, and as far south as Georgia and California, respectively. Infestations also occur in inland states (see Figure 13).

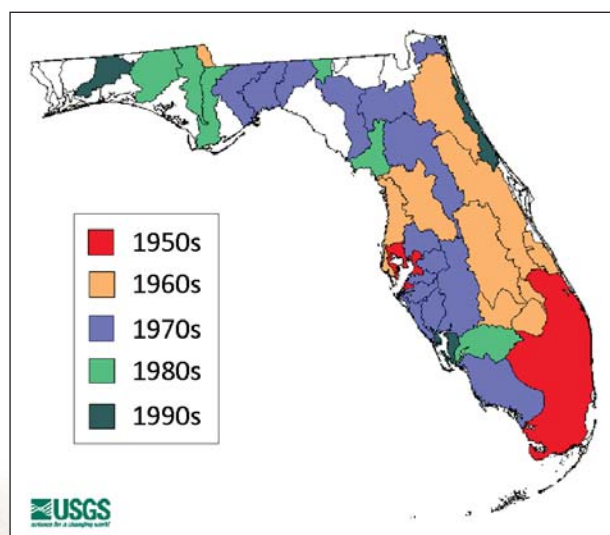


Figure 14. Spread of hydrilla in Florida over forty years, recorded from its introduction in the 1950s until its establishment in most counties in the 1990s. There is no data available on hydrilla introduction into the counties in white; it does not mean that there is no hydrilla present. Map created by the U.S. Geological Survey, Department of the Interior/USGS, available in the public domain.

Biotype — a form of the same plant species that shows special characters (for example, presence/absence of male or female flowers, resistance to a chemical herbicide, tolerance to extreme temperatures)

Dioecious — female and male flowers occur on different plants

Monoecious — female and male flowers occur on the same plant

Resistance [n.], resistant [adj.] (to herbicides) — the ability of a plant to survive the exposure to a typically lethal dose of herbicide

Invasive Properties of Hydrilla

There are several reasons why hydrilla is invasive. They define why this species is able to outcompete native aquatic weeds and how hydrilla has been able to spread so extensively.

TOLERANCE TO DIFFERENT HABITATS: Hydrilla thrives in a wide variety of conditions, from low to high temperature, low to high nutrient content, and low to high pH. It is even capable of growing in waters with salinity up to 7%.

LACK OF NATURAL ENEMIES: As hydrilla is exotic and not native to the U.S., it has no native natural enemies, so its growth can continue relatively unchecked.

FAST GROWTH: Each growing tip of hydrilla grows up to one inch per day. Once the plant starts to branch profusely near the water surface, the actual increase in biomass is exponential. In a recent experiment, a single nine inch long shoot produced over 3,200 inches of growth in five weeks.

COMPETITION FOR SUNLIGHT: Hydrilla is capable of growing in low-light conditions, down to 1% full sunlight. This allows hydrilla to start growing early in the year and for a long period each day, so it grows faster than other submersed plants. Once topped out, the plant blocks light from reaching other plants.

MULTIPLE METHODS OF PROPAGATION: Hydrilla uses multiple methods to propagate, including turions, tubers, fragments (Figure 15), and—where the monoecious type is present—seeds from flowers. Tubers and turions are very difficult to kill, and each tuber may grow into a plant that produces several thousand new tubers per square meter. Fragments are often taken from one water body to another on boating and fishing equipment. About 50% of fragments that have just one whorl of leaves will regenerate and form new plants (Figure 16).

Early Detection and Intervention Guidelines

If you think you have a new infestation of hydrilla or another invasive weed or animal that you have not seen before, you should **contact one of the state officials listed in the First Point of Contact in States with Hydrilla Infestations section** (page 126). These contacts should be able to help you find the correct point of contact for your situation.

What to Do When You Suspect a New Infestation

You have noticed (or somebody who called your office reported) increased growth of aquatic weeds on a shoreline? The weed looks like hydrilla or another species of invasive aquatic weeds? Here is what you do:

- Collect a sample of the weed to take or send to a specialist for identification. If this is not possible or permitted, then take photographs. It would also be helpful to record the GPS coordinates of the sample site.
- Call your local **Extension office** and report the problem.
- Call your state's **Fish and Wildlife Commission or Department of Environmental Protection** and have them connect you with an aquatic weed specialist.



Figure 15. Stem fragments of hydrilla with turions (top) and tubers (bottom). Photographs by Lyle Buss, University of Florida.

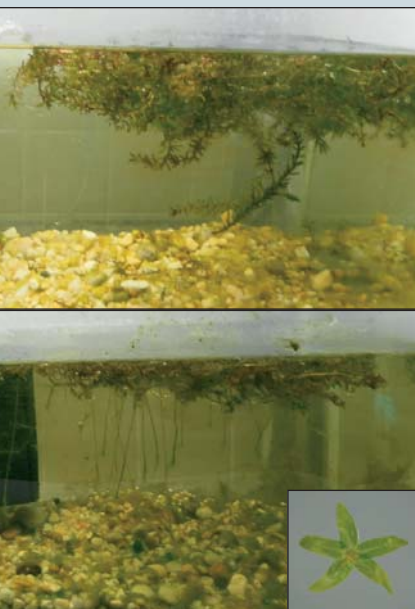


Figure 16. A fragment as small as one whorl of hydrilla (inset photo) can produce new plants. These fragments floating on the surface (top) form new roots that will anchor in the sediment (bottom). Photographs by Lyle Buss (inset photo) and Julie Baniszewski (top and bottom photos), University of Florida.

- Ask the specialist to assist you in identifying the weed and, if appropriate and necessary, in developing a management plan that will be in compliance with state and federal laws.

Remember, when you call the specialist to seek assistance with identification, **write down the mailing instructions you receive from the specialist** so that the sample will arrive intact and the specialist will be able to identify the plant.

Water body managers may have the resources and expertise to conduct an initial survey and provide the contacted specialist with details about the infestation. In the next three sections, we describe general initial steps that one can take when a new infestation occurs.

How to Survey for Hydrilla

Early detection under water may allow removal of the plants before they reach the surface and begin to form profusely branched, dense vegetation mats. In shallow areas, surveying the bottom from a boat with a viewing tube or from the water by snorkeling is possible. In deep waters, scuba diving might be necessary. An underwater video system is useful for scanning large areas.

For the survey, **space and locate transects by GPS** and make sure to include deep water areas as well as key points, such as boat ramps, swimming areas, intakes, and habitats of fish and birds.

Use ID cards or keys to identify the plants you record. If in doubt, take a plant sample and submit it for identification (see Contacts for Plant Identification and Management Advice on page 126). If you can, include the roots because hydrilla grows characteristic tubers that facilitate quick identification.

Most often, hydrilla enters a new habitat with water flow, boats, or aquatic birds. It is therefore important to survey areas such as inlets, upstream waters, access points, and known habitats of wading or shore birds.

How to Measure the Extent of an Infestation

Mapping the extent of a hydrilla infestation can be tricky. During early invasion, hydrilla disperses mainly by tubers and turions that remain in the sediment and will sprout in the following growth season.

When you discover hydrilla growth, follow up with visual inspection expanding in concentric circles from the center of growth. You may also follow the direction of the water current. In large water bodies, focus your attention on likely sources of invasion, such as inlets, boat ramps, docks, and bird habitats.

Habitat — an area or environment where a plant, fungus, or animal normally lives and grows

Monoecious — female and male flowers occur on the same plant

pH — a term used in chemistry to indicate the acidity or alkalinity of a solution based on the availability of free hydrogen; values range from 1 to 14; pH 7 = neutral, low pH (1 to <7) = acidic, high pH (>7 to 14) = alkaline

Submersed — a plant with most leaves growing underwater; flowers and some of the leaves may float on the water surface



Habitat — an area or environment where a plant, fungus, or animal normally lives and grows

Pesticide — a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals

Get a rough estimate of coverage, for example, the number of stems per area unit or the extent of the area covered by topped-out hydrilla. **Measuring and monitoring the extent of an invasion will help you determine if the applied control tactics are successful in reducing the infestation.**

There are several standardized techniques that have been developed for aquatic vegetation monitoring. Automatic electronic depth finders, which distinguish sediment from plant material, can be very useful to calculate the extent of an infestation. Whichever method you choose, **try to be as thorough as possible.** It is best to use a map of the water body; if none is available, draw one. Outline features of the shoreline and add reference points.

How to Report an Invasion in Florida

Once the presence of hydrilla is confirmed, contact the **Florida Fish and Wildlife Conservation Commission** to notify the town or county in which the infested water body is situated.

Also **contact your local UF/IFAS Extension Office.** You can find contact information on page 126 of this guide.

Are you Extension faculty? Here are actions you can take:

- Try to identify all stakeholder groups and contact as many as possible. These may include park staff, shoreline property owners, lake associations, birdwatchers, boaters, anglers, swimmers, and water suppliers, to name a few.
- Install educational signage at access points (docks, boat ramps, etc.) and write press releases for local media.
- Use local events to raise awareness in the public. People need to know what they can do to prevent the spread of invasive aquatic weeds. Many don't even know that hydrilla is invasive and causes environmental damage as well as economic losses.

Preventing New or Recurring Hydrilla Infestations

REMEMBER: PREVENTION COSTS LESS THAN TREATMENT!

Most freshwater bodies are suitable habitats for hydrilla because this invasive aquatic weed can tolerate a wide range of environmental conditions. Previously infested water bodies, of course, are already known to support hydrilla growth and should receive special attention.

A successful hydrilla education program needs to reach everyone who visits water bodies—in other words, people of all ages and interests. These include families with kids and dogs, boaters, anglers, hunters, water sport enthusiasts, water gardeners, park and lake managers, aquatic plant managers, and pesticide applicators.

HOW CAN YOU HELP PREVENT THE SPREAD OF HYDRILLA AND OTHER INVASIVE AQUATIC PLANTS AND ANIMALS?

- **Do not empty your aquarium contents into waterways** (Figure 17A). If you want to dispose of aquatic plants from your aquarium, place them onto a plastic sheet and allow them to dry out completely before you dispose of them in the trash.
- **Clean your boat, trailer, live wells, fishing equipment, and diving gear** before and after you visit a water body. Do this at the boat ramp, so that you won't forget later (Figure 17B+C).
- **Do not place any plant material back into the water.** Dispose of plant material in on-site trash cans or in your household trash. Do not compost hydrilla or any other aquatic weeds.
- Did your dog go swimming? **Please wash and brush your dog** thoroughly before allowing your pet to jump into new waters.
- **Be aware** that possession of hydrilla without a permit is illegal in Florida.

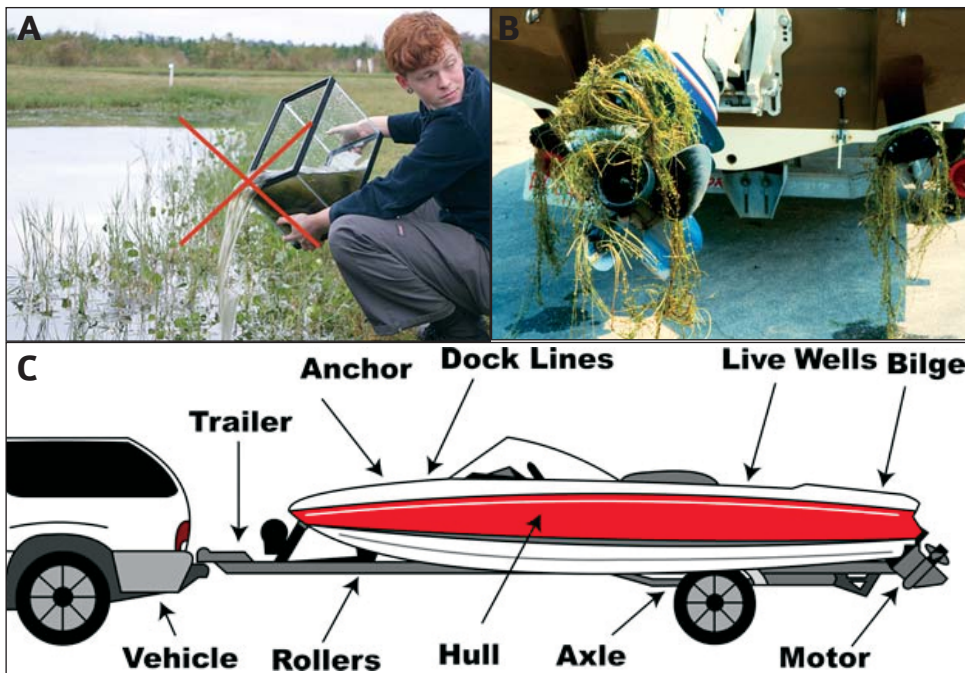


Figure 17. Prevent the spread of hydrilla. A. Please, no dumping! Once exotic plants from your aquarium are released into natural areas, the plants may establish and become invasive. This is what happened with hydrilla in many areas. B+C. Clean up! If hydrilla is tangled up on your motor (top photo), remove all plant material and dispose of it in the trash. Remember that each fragment can produce a whole new plant if released back into the water. Check and clean all areas indicated in the drawing (bottom). Photographs by unknown (A) and Don Schmitz (B), UF/IFAS Center for Aquatic and Invasive Plants. Drawing (C) by the California Department of Fish and Wildlife.



You can download the NOAA fact sheet from the public domain. URL: <http://www.habitat.noaa.gov/> (Scan the QR code to connect to the file.)



You can download the complete NANPCA of 1990 (PDF file, 45 pages) from the public domain. URL: <http://www.anstaskforce.gov/Documents/nanpca90.pdf> (Scan the QR code to connect to the file.)



You can download the complete NISA of 1996 (PDF file, 20 pages) from the public domain. URL: <http://www.anstaskforce.gov/Documents/NISA1996.pdf> (Scan the QR code to connect to the file.)

Federal Laws and Regulations

Hydrilla is a federally listed aquatic nuisance species. The federal regulations regarding the management of such species in the United States were enacted in 1990 and reauthorized in 1996 (see sections below). We retrieved the information in the following three subsections from a fact sheet published by the National Oceanic and Atmospheric Administration (NOAA) Fisheries Service.

Nonindigenous Aquatic Nuisance Prevention and Control Act (NANPCA) of 1990

The Nonindigenous Aquatic Nuisance Prevention and Control Act (NANPCA) was enacted in November 1990. Its main goals were: (1) to prevent the introduction and spread of invasive aquatic species into the waters of the United States; (2) to minimize the economic and ecological impacts caused by established nonindigenous aquatic species; and (3) to install a program that assists the states in the management and removal of nonindigenous aquatic species.

National Invasive Species Act (NISA) of 1996

In 1996, the National Invasive Species Act (NISA) reauthorized and amended the Nonindigenous Aquatic Nuisance Prevention and Control Act (NANPCA). Of great importance was the nationwide implementation of measures that would prevent the unintentional transport of established nuisance species to inland lakes and rivers within the United States. Two important pathways for such transport include recreational boat traffic and commercial barge traffic.

Aquatic Nuisance Species (ANS) Task Force

The Aquatic Nuisance Species (ANS) Task Force was established by the NANPCA and reauthorized by the NISA to coordinate activities among federal agencies and between federal agencies and organizations on the regional, state, tribal, and local levels.



More details are available at the website of the ANS Task Force. URL: <http://www.anstaskforce.gov/default.php> (Scan the QR code to connect to the website.)



Those of you who are interested in the current ANS Task Force Strategic Plan 2013-2017, please download the file (PDF, 29 pages) from the public domain. URL: <http://www.anstaskforce.gov/> (Scan the QR code to connect to the file.)

Chairpersons of the ANS Task Force are the director of the U.S. Fish and Wildlife Service and the undersecretary of Commerce for Oceans and Atmosphere. The membership includes 13 federal agencies and 12 ex-officio members. Regional panels, issue-specific committees, and work groups established by the ANS Task Force coordinate activities of the government with those of the private sector and with other North American interests.

State Laws and Regulations

Hydrilla verticillata

- Is on the **Federal Noxious Weed List** (U.S. Department of Agriculture, Animal and Plant Health Inspection Service, since the year 2000)
- Is on the **State List of Noxious Weeds** in Arizona, California, Florida, Maine, Mississippi, Nevada, New Hampshire, New Mexico, North Carolina, Oregon, South Carolina, Texas, Vermont, and Washington
- Is on the **State List of Prohibited Plant Species** in Indiana, Michigan, Nebraska, and Oklahoma
- Is on the **Florida Prohibited Plants** list, Florida Department of Environmental Protection
- Is on the **Florida Exotic Pest Plant Council (FLEPPC)** list of invasive plants
- Is, according to the FLEPPC, a **Category I plant**, which means it is “an invasive exotic plant that is altering native plant communities by displacing native species, changing community structures or ecological functions, or hybridizing with natives”

Florida State Law

Since July 2008, Florida’s invasive plant management program has been under the direction of the Florida Fish and Wildlife Conservation Commission (FWC).

The FWC determines the timing and the level of hydrilla management on each public water body after analyzing risks (addressing human health, economy, ecology, etc.), current water body conditions, primary uses, and available control technologies.

Florida law (F.S. 369.20) requires all persons intending to control or remove aquatic vegetation from waters of the state to obtain a permit from the FWC’s Invasive Plant Management Section unless an exemption for the activity has been provided in statute or rule (Chapter 68F-20).

Indigenous — occurring naturally in a place or region



Visit the UF/IFAS Center for Aquatic and Invasive Plants website, where you can download the above stated lists and access legislative chapters. URL: <http://plants.ifas.ufl.edu/node/634> (Scan the QR code to connect to the website.)



Application forms and laws for removing freshwater aquatic plants can be found at the Florida Fish and Wildlife Conservation Commission (FWC) website. URL: <http://myfwc.com/license/aquatic-plants> (Scan the QR code to connect to the website.)

Selected References (Early Detection)

Barko JW, Smart RM, McFarland DG, Chen RL. 1988. Interrelationships between the growth of *Hydrilla verticillata* (L.f.) Royle and sediment nutrient availability. *Aquatic Botany* 32: 205-216.

Glomski LM, Netherland MD. 2012. Does hydrilla grow an inch per day? Measuring short-term changes in shoot length to describe invasive potential. *Journal of Aquatic Plant Management* 50: 54-57.

Haller WT, Sutton DL, Barlowe WC. 1974. Effect of salinity on growth of several aquatic macrophytes. *Ecology* 55: 891-894.

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Chapter 4



Integrated Management Options

By Emma N.I. Weeks and Verena-U. Lietze

CONTENTS

Nutrient Management	28
Physical Control	32
Mechanical Control	36
Biological Control	39
Chemical Control	50

A variety of tactics can be used to manage hydrilla infestations. Please be aware that almost all types of management or control measures will require you to obtain a permit before you begin to manipulate the environment!

The next sections will give you detailed information on different approaches of hydrilla control that could be incorporated into an integrated pest management (IPM) plan. Some are very specific to hydrilla, whereas others are used to manage a variety of invasive aquatic plants.

Please Remember: Prevention costs less than treatment! See our Integrated Pest Management section that starts on page 4. Prevention should be the first step in any hydrilla IPM plan.

Nutrient Management

Excessive amounts of nutrients (especially nitrogen and phosphorus) will foster plant growth and may lead to an overabundance of aquatic vegetation. The major source of phosphorus is sediment that erodes from adjacent landscapes. Major sources of nitrogen are runoff from agricultural, turf, and sewage areas, atmospheric deposition, and leaching from septic tanks and sandy soils.

Monitoring these potential sources is important to ensure that they are not responsible for excessive nutrient loading into a water body that could be avoided. Updating aged septic tanks and maintaining stormwater treatment systems can be helpful in reducing nutrient loading. In agricultural areas, the use of fertilizers and irrigation systems can be planned to reduce runoff into neighboring water bodies.

In residential areas, similarly careful fertilization and irrigation of plants is helpful, but in addition, the type of vegetation surrounding the water body can help to protect it from runoff of nutrients. A study in Florida showed that increasing amounts of woody ornamental vegetation when compared to planting only turfgrass decreased the amount of nitrogen and phosphorus in the runoff.

When hydrilla gets out of control in a water body, it is usually only able to do so because of high nutrient levels, particularly nitrogen. Reducing the nutrient sources would be a more permanent way to reduce the ability of invasive plants like hydrilla to take over the water body. However, most water bodies in Florida naturally contain sufficient amounts of nutrient to facilitate plant growth and algal blooms without any external sources.

Keep this in mind when you evaluate a hydrilla infestation. Find out: Is there a possibility to reduce nutrient sources? Alternatively: Would it be feasible to change the nutrient content in the water?

Aluminum salts, iron salts, or calcium salts, when added to the water, bind to phosphorus in the water and the sediment and thereby immobilize phosphorus. This can be very effective for preventing or reducing the impact of algal blooms, which often occur following hydrilla control if there has been no effort to reduce nutrient sources.

Rooted vascular plants, such as hydrilla, are more limited by nitrogen than phosphorus. Unfortunately, there are no readily-available treatments for immobilization of excess nitrogen. To reduce the nitrogen load in water bodies, the best option is to improve management of runoff from nitrogen-rich areas. In the short-term, nitrogen-rich waters can be rerouted further downstream.

Nutrient management has been used successfully to control algae and improve water quality. However, removal of algae increases the clarity of water and thereby may facilitate the growth of weeds. **Consult with experts and learn more with the Green Industries Best Management Practices (GI-BMPs)** described in the next section.

Advantages and Disadvantages of Nutrient Management

Advantages: no hydrilla fragmentation, potentially not labor intensive, inexpensive

Disadvantages: not selective, does not remove hydrilla or kill tubers

Green Industries Best Management Practices (GI-BMPs)

The GI-BMPs are a science-based educational program for Green Industries workers (lawn-care and landscape-maintenance professionals), brought to you by the UF/IFAS Florida-Friendly Landscaping™ program. The GI-BMPs teach environmentally safe landscaping practices that help conserve and protect Florida's ground and surface waters. They can also save the Florida homeowner money, time, and effort; increase the beauty of the home landscape; and protect the health of your family, pets, and the environment.

This training is designed to provide corporate, governmental, environmental, and other personnel the Best Management Practices for lawn and landscape. Learn what impact the BMPs will have on your business or municipality. Developed by the Florida Department of Environmental Protection (FDEP) and endorsed by the pest control industry, this training is provided by the UF/IFAS Florida-Friendly Landscaping™ program with partial funding by the FDEP through a Nonpoint Source Management (Section 319h) grant from the U.S. Environmental Protection Agency.

Who Gets Trained in the GI-BMPs?

Florida Statute 482.1562 states that all current commercial fertilizer applicators must have a license from the Florida Department of Agriculture and Consumer Services (FDACS) as of January 1, 2014. To get this license, each Green Industries worker must be trained in the GI-BMPs and receive a certificate of completion from UF/IFAS and FDEP. Additionally, many non-commercial Green Industries applicators or other workers are required to pass the training by local ordinances or voluntarily participate in the program to better serve their clients.

Who Should Attend?

- Supervisors and employees in the lawn care, pest control, or landscape industries
- Municipal parks and recreation facility supervisors and employees
- Irrigation industry workers
- Commercial property managers
- City and county planning department staff
- City and county environmental department or water quality staff
- City and county commission staff

Pesticide — a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals



The information in this section is directly from the GI-BMP website (URL: <http://gibmp.ifas.ufl.edu>) and should be shared with people in your area if you believe fertilizers are being used improperly. (Scan the QR code to connect to the website.)

- Builders and developers
- Landscape designers and architects
- Golf course employees
- Anyone working in the areas of water quality, urban development, or environmental issues

Where can I find GI-BMP training classes?

In-person training classes are held at various UF/IFAS Extension offices around the state, industry offices, trade shows and association meetings, public buildings, and many other sites. For the training schedule, go to the URL: <http://gibmp.ifas.ufl.edu>

GI-BMP Online and DVD training also available:

- Industry professionals now have the option of taking the GI-BMP training online or using the free DVD training set.
- For online training information, go to the URL: <http://gibmp.ifas.ufl.edu>
- For DVD training information, go to the URL: <http://gibmp.ifas.ufl.edu>

Materials and Certification

Attendees will receive:

- Training in all major aspects of the GI-BMP manual
- GI-BMP manual (It is strongly recommended that you obtain and study this GI-BMP manual prior to attending class). Download the manual for free at the URL: <http://gibmp.ifas.ufl.edu> (on the top of the page, click the link “Click here to learn more about the GI-BMP program”)
- Certificate of completion (mailed 2 weeks after training—must pass post-test)
- Up to four Continuing Education Units (CEUs) for pesticide license holders—for in-person training only

How do I obtain my Limited Commercial Fertilizer Applicator Certificate (LCFAC)?

This certification, often referred to as the “state fertilizer license,” is issued by Florida Department of Agriculture and Consumer Services (FDACS).

- Apply online at the URL: <http://www.freshfromflorida.com>
- Download the LCFAC application form at the URL: <http://www.freshfromflorida.com>
- More LCFAC information from the FDACS is available online. URL: <http://www.freshfromflorida.com>

Selected References (Nutrient Management)

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Bellaud M. 2009. Cultural and physical control of aquatic weeds, pp 35-40. *In* Gettys LA, Haller WT, Bellaud M (editors). *Biology and control of aquatic plants: A best management practices handbook*. Aquatic Ecosystem Restoration Foundation, Marietta, Georgia.

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GI-BMP (Green Industry Best Management Practices). 2014. Training and program overview. URL: http://fyn.ifas.ufl.edu/professionals/BMP_overview.htm (23 July 2014).

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Stanley CD, Clarke RA, McNeal BL, Macleod BW. 2013. Impact of agricultural land use on nitrate levels in Lake Manatee, Florida. University of Florida, IFAS Extension. URL: <http://edis.ifas.ufl.edu/ss428> (8 May 2014).

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Figure 18. Manual removal of hydrilla by hand pulling. Photograph by UF/IFAS Center for Aquatic and Invasive Plants.



Figure 19. A diver performing suction harvesting. Photograph by UF/IFAS Center for Aquatic and Invasive Plants.

Physical Control

Physical control involves manipulating the physical environment in hydrilla-infested waters and may include the techniques described in the following sections. Bear in mind that most of the physical control methods (except for hand pulling and suction harvesting) are non-selective and affect all organisms in the manipulated area.

CAUTION: With many physical control methods, roots and tubers often stay behind and are a source for re-infestation, because hydrilla sprouts from tubers that can remain dormant in the sediment for several years.

Hand Pulling

Manual removal of plant material (Figure 18) brings success only when the entire plant, including roots and tubers, is removed. As the terms hand pulling and manual removal give away, this is real hands-on, labor-intensive work because you literally grab the hydrilla and pull it out of the water. This technique is particularly helpful in early infestations.

If hydrilla has spread to deep water areas, scuba divers will have to do this work. This takes time and may be expensive. Each diver removes around 90 plants per hour, and the cost is usually in the range of \$400 to \$1,000 per acre.

Advantages and Disadvantages of Hand Pulling

Advantages: selective, removes hydrilla

Disadvantages: unlikely to remove tubers, labor intensive, expensive, can cause fragmentation of plants

Suction Harvesting

This technique is particularly helpful in early infestations. The method is performed by divers who use hoses that are attached to a vacuum pump to selectively remove the target plant by the root system (Figure 19). The removed plant is deposited in a bag at the surface that traps the plant material but allows the water and sediment to drain back into the water body. Ideally, the divers manually pull hydrilla plants out of the sediment and so make sure that they do not leave behind tubers, which will sprout into new plants.

Like hand pulling, suction harvesting is labor intensive if done properly and involves specialized equipment. The costs are high, each acre may take up to a month to clear and cost from \$1,000 to \$25,000 depending on the speed of results. Suction harvesting is best followed by dredging to remove tubers and prevent regrowth. Dredging is a process during which mud, weeds, and other materials are scooped out of the bed of a water body (see page 34 for more details).

Advantages and Disadvantages of Suction Harvesting

Advantages: selective, removes hydrilla and tubers

Disadvantages: labor intensive, expensive, can cause fragmentation of plants

Surface Barriers

Surface barriers are non-selective options to keep areas of water bodies free of floating plant masses. Fences and booms often are used in areas created for swimming or for boat traffic. The plant material that accumulates within the barriers must be removed to prevent build-up of debris and release of nutrients into the water.

Fences usually consist of wire that is fixed to posts. The posts must project at least 2 feet above the mean annual water elevation of the waterway and must be marked with reflectors that can be seen from all directions of possible boat traffic. Booms consist of a floating barricade attached to a mesh skirt of variable depth (Figure 20). The mesh skirt allows water to flow but prevents plant movement.

Costs for installing surface barriers vary depending on the materials used and the amount of accumulated plant material that needs to be restricted.

Advantages and Disadvantages of Surface Barriers

Advantages: inexpensive, not labor intensive, no hydrilla fragmentation

Disadvantages: not selective, hydrilla not removed, tubers not killed

Benthic Barriers

Be advised that in Florida, it is illegal to cover large underwater areas, because it is possible that subterranean gas formations accumulate and may lead to dangerous eruptions.

Small patches of hydrilla populations may be controlled by covering the sediment with opaque fabric to exclude sunlight. Without sunlight, the plants cannot photosynthesize and will die. The fabric also acts like a weed blocker and provides a physical barrier to new growth (Figure 21).

Benthic barriers often are used in small ponds. Their use in areas with monocultures of invasive plants can be highly successful and can have minimal non-target effects. The barrier usually is made of plastic, fiberglass, nylon, burlap, or other non-toxic material. The barrier is rolled out from the shore and secured to the sediment with weights.

Professional installation costs in the range of \$10,000 to \$20,000 per acre. Although the barriers may last several seasons, they will need to be periodically cleaned. As always: Get expert help from your county's Extension office, your city, or the Fish and Wildlife Conservation Commission. Do not go out and try to solve the problem by yourself.

Advantages and Disadvantages of Benthic Barriers

Advantages: removes hydrilla and kills tubers, no hydrilla fragmentation

Disadvantages: not selective, labor intensive, expensive, possible gas buildup, illegal in some states

Monoculture — the agricultural practice of growing one crop on a farm or in a production area



Figure 20. Floating, curtain-style booms keep emergent aquatic vegetation contained. Photograph by UF/IFAS Center for Aquatic and Invasive Plants.



Figure 21. Benthic barrier (Aquascreen®) installed after a drawdown. Photograph by Matthew Vogt, New England Aquatic Services LLC.





Figure 22. Exposed and dried hydrilla during a lake drawdown. Photograph by Jeff Schardt, UF/IFAS Center for Aquatic and Invasive Plants.

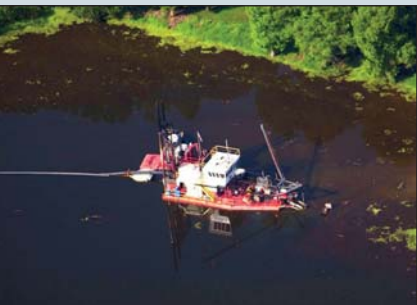


Figure 23. Dredging and pumping bottom sediments. Photograph by UF/IFAS Center for Aquatic and Invasive Plants.



Drawdowns

Lowering the water level is a possible control approach in water bodies with water level control structures. Without water, hydrilla plants dry out, die, and decompose (Figure 22). However, the exposure of the sediment to desiccation and extreme temperatures may also harm native aquatic plants and animals (such as frogs, turtles, mollusks, etc.).

In general, a period of 6-8 weeks is necessary to allow for full desiccation. Be aware that drawdowns will not kill vegetative propagules, such as turions and tubers. Following a drawdown, a plant species that uses turions and tubers to reproduce, for example hydrilla, may expand by taking advantage of the cleared area.

Drawdowns should be completed in the winter as it is easier to desiccate the plants in the absence of rain and high humidity. During winter, the risk is lower that the target plant may expand into deeper areas that cannot be fully drained. Unfortunately winter coincides with high wildlife and recreational use of water bodies in Florida.

To control hydrilla through drawdowns, these should be scheduled during winter and late summer. The first drawdown in the winter will kill existing plants and stimulate the sprouting of new plants in the sediment. The second drawdown in the late summer will kill these plants just before they can produce new tubers.

In most states, drawdowns must be authorized. Check with your state and local jurisdictions to find out about permits and other requirements.

Advantages and Disadvantages of Drawdowns

Advantages: inexpensive, not labor intensive, no hydrilla fragmentation

Disadvantages: hydrilla not removed, tubers not killed, not selective

Dredging

Dredging (Figure 23) is a process during which nutrient-rich sediment is removed from the bottom of a water body. As a result, nutrient-poor layers of the sediment are exposed and the depth of the water column is increased, thereby reducing the amount of nutrients available for plant growth and the amount of sunlight that reaches the bottom. For hydrilla, which is able to grow in very deep water, it would be unfeasible to excavate the water body to depths below the light compensation point. Hydrilla needs just 1% of sunlight to grow.

Dredging can impact the ecosystem due to the disturbance and removal of sediment. During dredging, aquatic organisms on or in the sediment may be damaged or removed. When sediment becomes suspended, light penetration is reduced, which reduces photosynthesis and consequently the release of oxygen by plants. Fish may be impacted when fine particles clog or damage their gills. As the particles settle on undisturbed sediment, they may smother benthic habitats or organisms.

To reduce non-target effects, use turbidity barriers or silt curtains around the site to limit the impacted area. The barrier or curtain is supported at the top through a boom-like floatation system and weighted at the bottom to ensure the suspended sediment is trapped. Additionally, changes to the sediment will alter the flow of a river or canal and may have other unpredicted impacts.

Depending on the water body, it may be necessary to get an environmental resource permit or a dredge and fill permit (in the Northwest Florida water management district) from the Florida Department of Environmental Protection or the appropriate Water Management District before you begin.

Advantages and Disadvantages of Dredging

Advantages: removes hydrilla and tubers, not labor intensive

Disadvantages: not selective, hydrilla fragmentation, expensive

Chaining

Chaining is a process where two tractors, one on either side of the water body, drag a chain across the bed of the water body. This method is particularly useful in canals that often have this type of access. As the chain moves along the sediment, plants are ripped up and float to the surface. As with other physical methods, care must be taken to remove all of the fragmented plant material.

Advantages and Disadvantages of Chaining

Advantages: inexpensive, not labor intensive, hydrilla removed

Disadvantages: not selective, tubers are not killed, fragmentation

Selected References (Physical Control)

Bellaud M. 2009. Cultural and physical control of aquatic weeds, pp 35-40. *In* Gettys LA, Haller WT, Bellaud M (editors). *Biology and control of aquatic plants: A best management practices handbook*. Aquatic Ecosystem Restoration Foundation, Marietta, Georgia.

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Mechanical Control

There are several types of machines that are utilized for aquatic plant management including cutters, shredders, rotovators, and harvesters. Harvesting, as the name suggests, results in removal of the plant material from the water body. Cutting, shredding, and rotovating involve the cutting of plant material into pieces that are too small to remain buoyant, and so they sink to the bottom. It is **not advisable to use methods that cut hydrilla into small pieces** as hydrilla is able to regenerate from fragments, so this method would increase the hydrilla problem. Mechanical harvesting can be very useful for hydrilla control and is described in detail below.

Mechanical Harvesting

Mechanical harvesting of hydrilla can be completed by specialized harvesters or by the combination of draglines or track hoes and disposal equipment (Figure 24).

Draglines and track hoes are large shovel machines. Draglines are cast using a cable system, and then the attached shovel scrapes plants and other material back to the shore. Track hoes are claw-like shovels that dig down into the sediment and pull plants back to shore. Both draglines and track hoes can be mounted onto a barge for offshore management.

In Florida, mechanical harvesting usually is performed by specialized machines (i.e., harvesters) that chop the hydrilla in large pieces and remove the cut hydrilla from the water and transport it to designated sites on shore for disposal and decomposition. Mechanical harvesters can operate in water bodies with a depth of at least one foot (about 30 cm) and have a cutting width from 5 to 12 feet (1.5 to 4.0 m). They operate relatively quickly creating an open area of one acre in approximately one hour.

Be aware that the machine is cutting the top of the plant only, the lower portions of the plant and roots are remaining in the water. Due to this, harvesting could be likened to mowing your lawn—it will have to be repeated before too long!

Caution: If hydrilla fragments are left behind in the water, they will produce new plants. In addition, the roots and tubers that are left behind are a source for re-infestation, because hydrilla sprouts from tubers that can remain dormant in the sediment for several years.

Considerations

One important consideration is the effect on non-target organisms. As well as not being selective with plants that are removed, mechanical harvesting can also kill animals that are within the harvested area. Examples of commonly killed animals are fish, crayfish, frogs, turtles, and snails. Juvenile sport fish have been shown to be particularly likely to get caught up in the removed plant mass. Studies have shown that 15-30% of some species can be removed from an area during a single harvest.



Figure 24. Mechanical removal by operating a harvester with removal belt (top) and by pushing weeds on shore for harvesting with a track hoe (bottom). Photographs by Jeff Schardt (top photo) and unknown (bottom photo), UF/IFAS Center for Aquatic and Invasive Plants.

Mechanical control may be considered when an infestation covers nearly the entire water body. During early colonization, however, fragmentation and incomplete removal during harvesting could further enhance the spread and growth rate of hydrilla.

Several companies have specialized in mechanical harvesting and should be consulted. To find them on the Internet, conduct a search using search terms like “aquatic mechanical harvesting,” “aquatic weed harvesting,” and “lake harvesters.” If you hire a company to harvest your plant material, make sure the machine is well cleaned and carefully inspected

before it is allowed to enter a new water body. Fragments of other invasive plants may be covering the machine and could be introduced if appropriate attention is not given to cleaning the machine between uses.

QUOTE: “I would like to know the results of the mechanical harvesting:

- How much is to be harvested
- How much was harvested-is it equal to what was scheduled to be harvested
- When is it scheduled to be harvested
- Where the harvested material will be dumped
- When it is dumped, is the site screened for nesting birds and other animals
- Most importantly, what else was harvested and how much...fish, snakes, turtles, birds, apple snails, mollusks, eel grass, Kissimmee grass, etc.”

— J.W.

Before you plan a mechanical harvesting project, contact your city or county to find out what permits are required and if mechanical harvesters would be allowed.

Be aware that residents in your area would like to be informed of hydrilla management plans and outcomes. Website reports and updates or newspaper articles are a great way to spread the word about what you are doing.

Advantages and Disadvantages of Mechanical Control

Advantages: removes hydrilla, not labor intensive

Disadvantages: not selective, expensive, fragmentation, tubers not killed

Selected References (Mechanical Control)

Haller WT. 2009. Mechanical control of aquatic weeds, pp 41-46. *In* Gettys LA, Haller WT, Bellaud M (editors). *Biology and control of aquatic plants: A best management practices handbook*. Aquatic Ecosystem Restoration Foundation, Marietta, Georgia.

McGehee JT. 1979. Mechanical hydrilla control in Orange Lake, Florida. *Journal of Aquatic Plant Management* 17: 58-61.

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Biological Control

Biological control is the intentional use of one organism to control or manage the growth of another organism.

A number of organisms that eat or infect hydrilla have been identified and have been or currently are being evaluated or used to keep hydrilla infestations in check. We call such organisms natural enemies. When natural enemies are used to manage, suppress, or eliminate invasive plants, people refer to them as biological control agents. You will find a list of all potential biological control agents of hydrilla on page 48.

Natural enemies of weeds can be herbivores (organisms that eat plants) or pathogens (organisms that cause diseases). Natural enemies of hydrilla that have been discovered include herbivorous fish, herbivorous insects, and a pathogenic fungus.

Biological control of aquatic weeds using herbivores requires that the herbivore has a preference for the target weed as a food source over other available food sources, with particular concern for native species.

There are two different approaches for biological control. Classical control involves the importation of a biological control agent. Usually the agent is identified during scouting missions to the native range of the invasive weed. Non-classical control involves either augmentative control, which is supplementing the natural populations of one or more natural enemies by mass release, or conservation control, which is protecting the natural enemy populations. Both of these techniques have been attempted for biological control of hydrilla.

Once identified, classical biological control agents are not instantly released. There are many steps that must be followed to ensure that the organism is safe before the release permit will be granted by the U.S. Department of Agriculture. These steps include host-range studies both in the native range of the invasive weed and in the laboratory under quarantine conditions on plants native to the U.S. Testing will be completed to determine how successful the organism is likely to be through climate tolerance studies and evaluation of hydrilla control efficacy.

Herbivorous Fish

The Asian grass carp (*Ctenopharyngodon idella*, Figure 25) is a non-specific yet effective consumer of hydrilla and other aquatic plants. Use of Asian grass carp to control hydrilla is classical biological control as these organisms were imported for that purpose.

Sterile grass carp have been used successfully to reduce hydrilla biomass in closed water systems. Because this fish is a non-native species, only sterile (triploid) grass carp can be released, and a permit is required in many states.

Contact your state's Fish and Wildlife Commission or a comparable regulatory agency about state-specific rules and regulations. Refer to the Contacts for Plant Identification and Management Advice section on page 126 to find a first point of contact in your state.

In Florida, for example, you will need to contact the Florida Fish and Wildlife Conservation Commission and request a permit for purchase and release of sterile grass carp. Refer to the Florida State Law section on page 25.

For more details on the Asian grass carp, check out the UF/IFAS Featured Creatures article in chapter 6.

Classical biological control

— the introduction of specialist natural enemies from the country of origin of a pest organism

Herbivorous [adj.], herbivory [n.]

— plant-eating

Pathogenic

— disease-causing

Triploid [adj.], triploidy [n.]

— when an organism has three sets of chromosomes; this is a rare condition in nature and leads to sterility in most organisms



Figure 25. Asian grass carp (*Ctenopharyngodon idella*).
Photograph by Ron Slay, Florida Fish Farms.





Get a permit for release of the Asian grass carp from the Florida Fish and Wildlife Conservation Commission (FWC). URL: <http://myfwc.com/wildlifehabitats/invasive-plants/grass-carp/> (Scan the QR code to connect to the website.)

Advantages and Disadvantages of Biological Control with Asian Grass Carp

Advantages: removes hydrilla preferentially, inexpensive, not labor intensive

Disadvantages: not selective, tubers not killed, hydrilla fragmentation

Steps for Using Asian Grass Carp in Florida

- 1. Identify your problem plant:** Ensure that it is one of the favorite foods of the Asian grass carp. The five most-preferred species in order of preference are hydrilla, musk grass (*Chara* spp.), southern naiad (*Najas guadalupensis*), Brazilian waterweed (*Egeria densa*) and watermeal (*Wolffia* spp.).
- 2. Determine the level of management you are looking for:** Asian grass carp will likely remove all plant material from the water body; in some cases (e.g., golf course ponds or canals) this may be desirable but in others this may be undesirable (e.g., ornamental ponds).
- 3. Get permission from any other users of the water body:** For example, property owners, and home owners associations.
- 4. Contact the Florida Fish and Wildlife Conservation Commission for a permit:** Asian grass carp release in Florida is restricted to permit holders only. You will fill out an application detailing the water body, which will be checked by a biologist. The biologist will recommend stocking rates, which will be a condition of your permit. More information is available from the Florida Fish and Wildlife Conservation Commission's website. URL: <http://myfwc.com/wildlifehabitats/invasive-plants/grass-carp/>
- 5. Install barriers to any escape routes:** The biologist will identify potential escape routes, which need to be fitted with barriers before fish are released. Barriers should have a maximum gap of 1.5 inches (provided the stocked fish are greater than 10 inches in length). As the applicant, you are required to get approval for and subsequently to maintain these barriers. More information on barrier construction is available from the Florida Fish and Wildlife Conservation Commission's website (see above).
- 6. Identify a supplier:** The Florida Fish and Wildlife Conservation Commission will provide a list of certified suppliers. In 2014, there were 42 certified suppliers, and 38 of them are located in Florida. You must use one of these suppliers if you release Asian grass carp in Florida.

Stocking Rates and Costs

The Florida Fish and Wildlife Conservation Commission typically recommends between three and ten Asian grass carp per acre, and each fish costs between \$5 and \$15. Therefore, costs can be as low as \$15 per acre.

When stocking with Asian grass carp, consider that they will eventually need to be removed once control of the aquatic weed has been achieved. Removal is not easy (without killing all fish in the water body) and requires a permit.

Asian grass carp are generally only permitted to be stocked into closed water bodies; in open water bodies, any canals or channels leading into other areas must be blocked with barriers to prevent fish escape. The barriers need to have a fine-enough mesh to prevent the smallest fish swimming through and must be high enough so that the fish cannot jump over them.

Sterility

Sterile Asian grass carp were developed by subjecting eggs to stress—either heat stress (hot or cold) or pressure. The stress causes the egg to retain an extra set of chromosomes and become triploid instead of diploid. Triploid individuals cannot reproduce as the presence of three chromosomes disrupts meiosis and prevents the production of viable eggs and sperm.

In triploid female fish, egg production does not occur, but in triploid male fish, sperm production can be induced. However, offspring from such triploid males were malformed, most died within one week, and the remainder died within a month. So although physiologically the males are not sterile, triploid grass carp are functionally sterile.

Concerns over the success rate of the sterilization technique have led to screening for diploid individuals by measuring the diameter of cell nuclei, as triploid individuals have larger nuclei. Blood samples are taken from every single fish that is to be sold as a certified triploid grass carp, and the sample is tested by the owner of the company (Figure 26) to ensure that the diameter of the cell nuclei is large enough to indicate the amount of DNA that would be present if the organism had three sets of chromosomes and is therefore sterile.

Additionally, the U.S. Fish and Wildlife Service visit the facility and test a randomly selected sample of each lot to verify the triploidy and sterility of the lot to be released. If a single diploid fish is found in the sample, the owner must retest the whole lot.



Figure 26. Processing blood samples from Asian grass carp to test for sterility. Photograph by Ron Slay, Florida Fish Farms.

Diploid — a cell with two sets of chromosomes, usually one set is from the organism's mother and one set is from the organism's father; diploid cells are the building blocks for meiosis (see below for definition)

Meiosis — when a diploid cell divides into haploid cells to produce reproductive gametes (this is the process required to produce sperm and eggs or pollen and ovules)

Nucleus (cell nucleus) [sing.], nuclei [pl.] — a membrane-bound organelle found in most cells; this organelle contains most of a cell's genetic material, and it is where the chromosomes are located

Triploid [adj.], triploidy [n.] — when an organism has three sets of chromosomes; this is a rare condition in nature and leads to sterility in most organisms

Herbicide — a substance that kills weeds (usually a chemical compound)

Triploid [adj.], triploidy [n.] — when an organism has three sets of chromosomes; this is a rare condition in nature and leads to sterility in most organisms

Plant species consumed by the Asian grass carp (in order of preference):

1. Hydrilla
2. Muskgrass
3. Southern Waternymph / Southern Naiad
4. Brazilian Waterweed
5. Watermeal
6. Duckweed
7. Azolla / Waterfern / Mosquitofern
8. Pondweeds
9. Coontail
10. Torpedograss

Considerations

Birds, snakes, otters, and other species of fish may prey on small Asian grass carp. Largemouth bass (*Micropterus salmoides* Lacepede) are a particular problem, and in their presence, any Asian grass carp smaller than 18 inches (45 cm) may be consumed. Therefore, if largemouth bass are present, stock fish that are larger than 12 inches (30 cm) or 1 lb (0.45 kg). If the plant material is dense, smaller fish will have increased chance of survival, so if fish are added following chemical or mechanical control, then one should consider stocking with larger fish.

In order that hydrilla consumption by the fish exceeds the growth rate of the plant, several factors need to be considered including age and sex of the fish. Depending upon these factors and the abundance and location of the plants within the water body, a stocking density needs to be selected (as done during the management described in Figures 27 and 28). Water quality factors may also play a role—particularly low dissolved oxygen (less than 3 ppm) and high temperatures (greater than 85°F or 27.8°C) may reduce management success.

In water bodies with such problems, it would be necessary to remove some of the weed biomass before stocking with fish. If herbicides or mechanical harvesting are used, it is necessary to wait before stocking with fish, because when hydrilla is killed with herbicide or removed by a harvester, the oxygen level in the water is likely to drop. It is important for fish survival that the oxygen level is allowed to stabilize (above 3 ppm minimum) before fish are introduced into the water body. After removal of some of the hydrilla biomass, lower stocking rates of carp will be needed to achieve management.

When Asian grass carp is used as part of an integrated pest management program, most other tactics should be completed before stocking with fish so that the fish are not effected by subsequent control efforts. After application of herbicides containing amine endothall or copper, which may have a negative effect on fish at the label rates, a waiting period will allow the ingredients to degrade sufficiently before the carp are added to the water body. It is also advisable to stock during colder months when fish are more likely to acclimatize well and less likely to contract diseases.

An ecosystem that has been supplemented with grass carp will change in several ways if the aquatic vegetation is eliminated. Firstly, growth of algae and abundance of phytoplankton will increase causing a decrease in water clarity. Fish species that are reliant on vegetation (e.g., chain pickerel, bluespotted sunfish, and golden topminnow) will decline and those that feed on phytoplankton (e.g., gizzard shad and threadfin shad) will increase in number. This has occurred in several lakes in Florida that were stocked with grass carp.

When using Asian grass carp, be aware that once the hydrilla is under control, if the fish are not removed, they will start to eat other less preferred but still highly palatable species. The top ten most preferred plant species for Asian grass carp are listed to the left (extracted from Sutton et al. 2012).

Remember: A permit is required for use, possession, and removal of Asian grass carp, and only certified triploid Asian grass carp may be used for management of aquatic weeds in Florida.



Figure 27. A pond in southeast Florida with a heavy infestation by hydrilla before management (left) and one year after stocking the pond with Asian grass carp at 40 carp per acre (right). Photographs by David Sutton (retired), University of Florida.



Figure 28. A citrus irrigation and drainage ditch in southwest Florida with a heavy infestation by hydrilla before management (left) and after herbicide treatment followed up by stocking with Asian grass carp at 20 carp per acre (right). Photographs by David Sutton (retired), University of Florida.

Herbivorous Insects

In addition to fish, insects also can be efficient consumers of plant material. Of the insects found associated with hydrilla, six species have been assessed for their impact on hydrilla infestations. You will find a brief description of these insects on the next few pages. For details and more photos of these insects and several others that have been described to occur on hydrilla in Florida, read the UF/IFAS Featured Creatures articles in chapter 6.

In the 1970s, researchers began searching for herbivorous aquatic insects that would consume hydrilla in amounts sufficient to reduce hydrilla biomass. They identified a number of promising insects, which have been or are being tested for their potential as biological control agents for hydrilla. In the 1980s, several insect species that specifically feed on hydrilla were introduced and released in the U.S. as classical biological control, these include two species of weevils and two species of flies.

Classical biological control

— the introduction of specialist natural enemies from the country of origin of a pest organism

Herbivorous [adj.], herbivory

[n.] — plant-eating

Classical biological control

— the introduction of specialist natural enemies from the country of origin of a pest organism

Habitat — an area or environment where a plant, fungus, or animal normally lives and grows

Herbivorous [adj.], herbivory [n.] — plant-eating

Submersed — a plant with most leaves growing underwater; flowers and some of the leaves may float on the water surface

Terrestrial — relating to the earth, for example, an organism that lives on land (as opposed to one that lives in water)



Figure 29. Larvae (top) and adult (bottom) of the hydrilla stem weevil, *Bagous hydrillae*. Photographs by Gary Buckingham, U.S. Department of Agriculture, Agricultural Research Service.

There also have been several species that have been identified as potential agents for non-classical control through augmentation or mass rearing of an organism and release to supplement the wild populations. These two organisms, one moth and one non-biting midge, are believed to be not native but they were not deliberately introduced for hydrilla biological control. Their route of arrival to the U.S. is unknown, but they were most likely introduced along with hydrilla.

Each organism will be discussed below in some detail, but if you would like more information on their biology, please review the UF/IFAS Featured Creatures articles in chapter 6.

Advantages and Disadvantages of Biological Control with Herbivorous Insects

Advantages: inexpensive, not labor intensive, selective, hydrilla removed in laboratory tests

Disadvantages: tubers usually not killed, hydrilla fragmentation

Classical Biological Control Agents

Classical biological control involves the importation of a biological control agent, usually a natural enemy from the native range of the invasive plant.

HYDRILLA STEM WEEVIL

The hydrilla stem weevil, *Bagous hydrillae* O'Brien (Insecta: Coleoptera: Curculionidae), is a semi-aquatic weevil that feeds on submersed hydrilla during both the larval and adult life stages (Figure 29).

The hydrilla stem weevil larvae feed within the stems, and the adults feed on the leaves and around the leaf nodes. The hydrilla stem weevil is native to Australia where it was collected in 1982. The weevil was introduced into water bodies in the U.S. in 1991. Unfortunately, the hydrilla stem weevil has not established in Florida. It is believed that the reason lies in the life cycle.

The hydrilla stem weevil larvae require a terrestrial habitat to pupate. In the native range, hydrilla mats break off and drift to the side, and the larvae pupate in the stranded plant material on the bank or directly in the silt of the bank itself. As accumulation of plant material at the edge of the water body rarely occurs in Florida, the life cycle of this insect cannot be completed.

For more details on the hydrilla stem weevil, check out the UF/IFAS Featured Creatures article in chapter 6.

HYDRILLA TUBER WEEVIL

The hydrilla tuber weevil, *Bagous affinis* Hustache (Insecta: Coleoptera: Curculionidae), is a semi-aquatic weevil (Figure 30). The larvae and the adults of this species feed on hydrilla.

The hydrilla tuber weevil larvae feed on the tubers, and the adults feed on tubers, stems, and leaves. The hydrilla tuber weevil is native to India, where it was collected in 1982, and was introduced into U.S. water bodies in 1987. Unfortunately, like the hydrilla stem weevil, the hydrilla tuber weevil has not established in Florida. Lack of establishment is believed to be due to it not being possible for the weevil to complete its life cycle in Florida.

The hydrilla tuber weevil adults lay their eggs into stranded hydrilla or the soil as the water retreats in the dry season. The larvae hatch and migrate to tubers where they feed. The larvae of this species pupate within the tuber or in the surrounding soil. Exposed tubers are rare in Florida so this species is unable to complete its life cycle.

For more details on the hydrilla tuber weevil, check out the UF/IFAS Featured Creatures article in chapter 6.

HYDRILLA LEAF-MINING FLIES

The hydrilla leaf-mining flies or *Hydrellia* species flies are another group of hydrilla-eating insects (Figure 31). *Hydrellia* species flies feed on hydrilla during the larval stage. The larvae are leaf miners; they burrow into the leaves and feed on the inner tissue. Feeding results in transparent leaves, which reduces the photosynthetic capacity of the plant. The mined leaves are also less buoyant, and the plant will sink and die if heavily infested.

There are four species in Florida, two native and two introduced. The two introduced species are the Asian hydrilla leaf-mining fly, *Hydrellia pakistanae* Deonier, and the Australian hydrilla leaf-mining fly, *Hydrellia balciunasi* Bock (Insecta: Diptera: Ephydriidae). The species with the potential to have the highest impact on hydrilla is the Asian hydrilla leaf-mining fly, *Hydrellia pakistanae*. It was introduced to Florida after being collected in India. Its native range includes India, Pakistan, and China. The fly has dispersed, and to date, most hydrilla-infested water bodies support populations of *Hydrellia pakistanae*.

Although this fly is found everywhere that hydrilla is a problem, it does not seem to be assisting in management. One of the reasons given for this lack of impact success is that the populations stay low, perhaps due to parasitism by the hydrellia fly parasitic wasp, *Trichopria columbiana* (Ashmead) (Insecta: Hymenoptera: Diapriidae).

For more details on the hydrilla leaf-mining fly and the hydrellia fly parasitic wasp, check out the UF/IFAS Featured Creatures articles in chapter 6.

Non-Classical Biological Control Agents

Non-classical biological control involves supplementing native or naturalized populations of insects, or conservation of insect populations to increase numbers.

HYDRILLA TIP MINING MIDGE

The hydrilla tip mining midge, *Cricotopus lebetis* Sublette (Insecta: Diptera: Chironomidae, Figure 32), is another promising hydrilla control candidate. Hydrilla tip mining midge larvae feed and develop inside growing stem tips and cause the damaged tips to break off and decompose. Extensive herbivory would prevent growth of the hydrilla and the formation of hydrilla surface mats. The adults are non-feeding (and therefore non-biting) and short-lived.



Figure 30. Larva (top) and adults (bottom) of the hydrilla tuber weevil, *Bagous affinis*. Photographs by U.S. Department of Agriculture, Agricultural Research Service.



Figure 31. Larva (top) and adult (bottom) of the hydrilla leaf-mining fly, *Hydrellia* spp. Photographs by Lyle Buss, University of Florida.



Figure 33. Larva (with eggs; top) and adult (bottom) of the hydrilla leafcutter moth, *Parapoynx diminutalis*. Photographs by Lyle Buss, University of Florida.



Figure 34. Larva (top) and adult (bottom) of the waterlily leafcutter moth, *Elophila oblitalis*. Photographs by James Cuda (top) and Lyle Buss (bottom), University of Florida.

Unlike the previous three species, the hydrilla tip mining midge was discovered in Florida. U.S. Department of Agriculture researchers identified the midge from hydrilla exhibiting stunted growth in Kings Bay, Crystal River. It is not known how the midge arrived in the U.S., but this insect is not believed to be native. It was most likely introduced along with hydrilla by the aquarium trade.

The UF/IFAS Hydrilla IPM RAMP research team currently is evaluating the control potential of the hydrilla tip mining midge in an integrated approach when it is combined with other management tactics. Results of our research are being published and studies are still underway.

For more details on the hydrilla tip mining midge, check out the UF/IFAS Featured Creatures article in chapter 6. For more details on the research of the UF/IFAS Hydrilla IPM RAMP team on integrated methods with the hydrilla tip mining midge see pages 66 and 67. Future results and updated information will be posted on the UF/IFAS Hydrilla IPM RAMP website (see page 47).



Figure 32. Egg mass, larva, and female adult of the hydrilla tip mining midge, *Cricotopus lebetis*. Photographs by Jerry Butler (retired), University of Florida.

HYDRILLA LEAFCUTTER MOTH

The hydrilla leafcutter moth, *Parapoynx diminutalis* Snellen (Insecta: Lepidoptera: Crambidae, Figure 33), also was found feeding on hydrilla in Florida. The larvae of the moth feed on hydrilla leaves and stems and use plant material to construct cocoons. Although the main food source for the hydrilla leafcutter moth is hydrilla, the larvae are not host specific and will complete their development on several other aquatic plants.

Although the moth was originally discovered in Florida in 1976, its native range is in Asia, Africa, and Australia. Prior to this discovery, the moth had been identified during scouting trips to India and Pakistan in 1971 and considered as a potential biological control agent for hydrilla. However, due to its generalist feeding, an importation permit was not granted. The route of arrival into the U.S. of the moth is unknown, but the moth is believed to have arrived with hydrilla via the aquarium industry.

Another closely related moth that frequently feeds on hydrilla is the waterlily leafcutter moth, *Elophila oblitalis* (Walker) (Insecta: Lepidoptera: Crambidae, Figure 34). Like the larvae of the hydrilla leafcutter moth, the larvae of this species feed on hydrilla but are even less specific than the hydrilla leafcutter moth.

For more details on the hydrilla leafcutter moth and the waterlily leafcutter moth, check out the UF/IFAS Featured Creatures articles in chapter 6.

Pathogenic Fungus

As mentioned before, natural enemies of plants include not only animals that would feed on them but also microorganisms that can cause disease. The fungus *Mycoleptodiscus terrestris* (Mt) is a pathogen of hydrilla. It was isolated first in 1987 from hydrilla growing in different parts of the U.S. and subsequently formulated and tested as a biological control agent. The fungal inoculum operates much like a chemical herbicide in that it contacts, penetrates, and kills hydrilla.

The infection occurs 8-24 hours after application. Within 4-7 days, the plant will begin to yellow, and by 7-14 days, the upper plant material will begin to disintegrate. Although the entire plant is not killed by the treatment, the fungus provides short-term to intermediate control of the vegetative mats that cause the greatest issue when hydrilla is topped out.

Under various experimental conditions, Mt fungus has significantly reduced hydrilla biomass when applied alone or in combination with chemical herbicides (Figure 35). When Mt fungus was combined with fluridone, hydrilla biomass was reduced by 93% compared to an untreated control. Either treatment alone achieved a maximum biomass reduction of 40%.

This difference shows that the two control methods can be combined to give a synergistic effect, meaning that the effect of the two methods together is greater than the sum of the effect of the two methods. Similar results were observed when Mt fungus was combined with the herbicide endothall.

Currently, the Mt fungus cannot be released into Florida water bodies. A permit needs to be obtained to import the fungus. However, at present (2014), the Mt fungus is not commercially available. Hydrilla IPM RAMP researchers now are testing its compatibility with the hydrilla tip mining midge. Read more about our results on page 66.

Advantages and Disadvantages of Biological Control with Pathogenic Fungus

Advantages: no non-target toxicity, species specific, works well when integrated with other methods

Disadvantages: slow acting, low success rate when used alone



Visit the UF/IFAS Hydrilla IPM RAMP website for our most up-to-date recommendations.
URL: <http://entomology.ifas.ufl.edu/hydrilla>
(Scan the QR code to connect to the website.)

Herbicide — a substance that kills weeds (usually a chemical compound)

Indigenous — occurring naturally in a place or region

Pathogenic — disease-causing



Figure 35. Hydrilla growth following no treatment (top) versus combined treatment with a chemical herbicide and the plant pathogenic fungus *Mycoleptodiscus terrestris* (Mt) (bottom). Photographs by Judy Shearer, U.S. Army Corps of Engineers.

Summary Table: Potential Biological Control Agents of Hydrilla

Common name	Type	Native range	Introduced	Established	Host specific	Damages	Pages
Hydrilla stem weevil	Classical	Australia	1991	Maybe	Yes	Stems Leaves	44, 101
Hydrilla tuber weevil	Classical	India Pakistan	1987	No	Yes	Tubers Stems Leaves	45, 106
Hydrilla leafcutter moth	Non-classical	Asia Africa Australia	Unknown	Yes	No	Leaves Stems	46, 78
Hydrilla tip mining midge	Non-classical	Unknown	Unknown	Yes	Yes	Growing stem tips	45, 96
Asian hydrilla leaf-mining fly	Classical	Asia	1987	Yes	Yes	Leaves	45, 83
Australian hydrilla leaf-mining fly	Classical	Australia	1989	No	Yes	Leaves	45, 83
Mt fungus	Non-classical	United States	Not applicable	Not applicable	Yes	Leaves Stems	47
Asian grass carp	Classical	Russia China	1970	Yes	No	Leaves Stems	39, 117

Selected References (Biological Control)

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Habitat — an area or environment where a plant, fungus, or animal normally lives and grows



Use this search engine to find out if a product is registered by the Environmental Protection Agency (EPA). URL: <http://iaspub.epa.gov/apex/pesticides/f?p=PPLS:1>
(Scan the QR code to connect to the search engine.)



Use this search engine provided by the Florida Department of Agriculture and Consumer Services (FDACS) to find out if a product is licensed for use in Florida. URL: <http://www.freshfromflorida.com/Divisions-Offices/Agricultural-Environmental-Services/Business-Services/Pesticide-Brand-Registration>
(Scan the QR code to connect to the search engine.)

Chemical (Herbicide) Control

An herbicide is a type of pesticide that is used for killing plants, usually weeds. An aquatic herbicide is used in aquatic ecosystems for the control of aquatic plants that are having an adverse effect on the environment. Always check to be sure an herbicide is still legal for use in your area before you make an application.

Advantages and Disadvantages of Chemical Control

Advantages: removes hydrilla, inexpensive, not labor intensive, no fragmentation of plants

Disadvantages: often not selective without selective use, usually does not kill tubers

History of Aquatic Herbicide Use

Although aquatic herbicides have been used since the late 1880s to control invasive aquatic plants, the majority of management was implemented by mechanical and physical efforts. Unfortunately, these methods alone were not able to keep up with the invasive properties of aquatic weeds, and in 1902, an act was passed by U.S. congress that permitted the use of mechanical, chemical or any other means for extermination of the weeds. Several methods were used, of these only copper remains in use to this day.

New herbicides began to be developed in the 1940s, and by 1975, around 500 new pesticides had been discovered. The broad-scale application of herbicides to environmentally sensitive areas led to public and professional concern. Consequently, the Environmental Protection Agency (EPA) was formed in 1970 to regulate pesticide registration and use to protect humans and the environment from potential side-effects of pesticide applications. Presently, due to concerns of pesticide use and resistance, integrated pest management programs are being designed to ensure sustainable control.

Currently, there are fourteen active ingredients that are registered for use in Florida waterways by the EPA and the Florida Department of Agriculture and Consumer Services (FDACS). Eight of these are labeled as specifically targeting hydrilla. Of these, six were used in 2013 for control of hydrilla in Florida. See the Summary Table on page 53 for a breakdown of these chemicals and their uses in Florida. Further information about each of the eight active ingredients is provided below.

Depending on the habitat and vegetation, several active ingredients have the potential to control hydrilla selectively—in other words, to kill hydrilla while not affecting other aquatic plant species.

In some situations certain herbicides may not be permitted for use. Before using herbicides, make sure that your aquatic applicator license is up-to-date, that you have permission from local authorities to apply an herbicide to a water body, that you have read the label (see pages 57-61), and that you follow the recommendations for personal protective equipment (PPE), and application rate and time!

Herbicides Approved for Aquatic Use and Applied in Florida

The aquatic herbicide that is used will depend on the type of water body and extent of the problem. It is important to consider non-target effects of each herbicide on animals and plants. In sensitive areas, such as conservation areas, it is not desirable to apply a broad-spectrum herbicide that is toxic to invertebrates if an alternative is available. Broad-spectrum herbicides may kill many species of plants and potentially insects and other animals and—especially if used incorrectly—may have a negative impact on the ecosystem.

There are several important things to consider when choosing an herbicide:

- Contact or systemic (see below for description)
- Selectivity for target plant
- Toxicity to animals
- Speed of control
- Duration of control

When you have selected your appropriate herbicide, it is important to consider timing of the application. Timing is crucial for safe and effective hydrilla control. In Florida, control during the summer months is not advisable as the dissolved oxygen content of the water is often so low already that the ecosystem could not handle dead plant material that would further reduce the oxygen levels. Treatment during Florida's rainy season is not advised as high variation in the water level can dilute concentrations of herbicide and reduce effectiveness.

In the spring in Florida, native plants begin to grow and become more susceptible to herbicide treatments. Hydrilla continues to grow later in the fall and begins to grow earlier in the spring than most native plants, and this growth habit is part of the reason why it has such an advantage over native plants. This is the window of opportunity when herbicide treatments can be selective towards hydrilla. In addition to these factors, you may need to consider the other users of the water body and their timing requirements (for example, wildlife, recreational activities, and irrigation needs).

Remember: A state permit may be required prior to herbicide application depending on the area being treated. Contact the Florida Department of Agriculture and Consumer Services (FDACS) and the Florida Fish and Wildlife Conservation Commission (FWC) before making any applications of pesticide to get advice on effective treatments and permits that may be necessary.

Contact or Systemic

Aquatic herbicides may have contact or systemic action. **Contact herbicides** cause injury to exposed plant tissue and usually are fast acting. However, as the herbicide does not move through the plant, only exposed tissue is damaged. Regrowth may occur relatively quickly from undamaged plant material or protected roots or rhizomes. A negative impact of the fast kill by contact herbicides is the rapid drop in oxygen caused by decaying plant tissue, which may cause fish mortality. An example of a contact herbicide currently used for hydrilla control in Florida is endothall (2014).

Systemic herbicides are absorbed into the plant tissue and then moved throughout the plant to the site of action. Systemic herbicide treatments will have an effect on plant tissue that is not directly treated. Systemic herbicides are slower acting than contact herbicides. One benefit of slower action is that there is a less dramatic effect on oxygen levels as the dying plants decay, which improves fish survival. An example of a systemic herbicide currently used for hydrilla control in Florida is fluridone (2014).

Herbicide — a substance that kills weeds (usually a chemical compound)

Monoculture — the agricultural practice of growing one crop on a farm or in a production area

Pesticide — a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals



Visit the Florida Department of Agriculture and Consumer Services (FDACS) online. URL: <http://www.freshfromflorida.com>
(Scan the QR code to connect to the website.)



Visit the Florida Fish and Wildlife Conservation Commission (FWC) online. URL: <http://myfwc.com/license/aquatic-plants>
(Scan the QR code to connect to the website.)

Herbicide — a substance that kills weeds (usually a chemical compound)

Pesticide — a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals



Pesticide safety information from the UF/IFAS Electronic Data Information Source (EDIS) is available online. URL: http://edis.ifas.ufl.edu/topic_pesticide_safety (Scan the QR code to connect to the website.)

Selectivity for Target Plant

Some herbicides are selective in their actions, although many are not and have broad-spectrum effects on many species of plants. The selectivity of broader-spectrum herbicides can be increased by well-planned applications. Several factors can influence the selectivity of an herbicide including timing of the application, method of application, and concentration. Treating during the winter and spring will provide selective control of hydrilla as most other plants are dormant during this time.

Selectively treating only monocultures of hydrilla with broad-scale application methods, such as aerial spraying, will reduce effects on non-target plants. If the hydrilla is mixed with native non-target plants, then spot applications will reduce effects on desirable species. Concentration also can be used to alter selectivity. Applying low concentrations of herbicides for longer periods of time often provides selective control of hydrilla and protects native vegetation.

Toxicity to Animals

Most herbicides that demonstrate any level of toxicity to animals are restricted in use; an example is copper, which is toxic to animals when applied at incorrect concentrations. During the registration process for aquatic herbicides, the U.S. Environmental Protection Agency (EPA) takes great care to issue permits for only those active ingredients or formulations that will not harm animals. Furthermore, aquatic plant managers are advised to use the least toxic of the herbicides suitable. For example, herbicides that act on the acetolactate synthase pathway are specific to plants as animals lack this enzyme. Herbicides that act on this pathway are the systemic herbicides bispyribac-sodium, imazamox, and penoxsulam.

Speed of Control

Contact herbicides in general are faster acting than systemic herbicides. Systemic herbicides need time to move through the plant to the sites of action. Although in many instances a quick fix is desirable, this can lead to additional problems. If a lot of plants die quickly, then the oxygen level in the water drops, which can have non-target effects particularly on fish. If contact herbicides are to be used on a large area with high plant density, it may be desirable to remove some of the vegetation first through mechanical harvesting or other methods.

Duration of Control

Although contact herbicides act quicker, systemic herbicides usually have a longer duration of control. This is because more plant tissue is damaged and because the tubers and turions are sometimes killed. Duration of control is important as increased applications of herbicide cost more money, involve more labor, and lead to more chemical input into the ecosystem.

Herbicides Used for Hydrilla Control in Florida

Eight of the herbicides approved for aquatic use in the United States are being used currently in Florida for hydrilla control (see Summary Table in the next section). The following sections provide an overview of their modes of action and the advantages/disadvantages associated with their use.

A number of helpful documents are available for you if you need to familiarize yourself with the safe use of pesticides. Find what you need at the UF/IFAS Electronic Data Information Source (EDIS).

SUMMARY TABLE: AQUATIC HERBICIDES APPROVED FOR USE IN PUBLIC WATERS IN FLORIDA

This table lists the aquatic herbicides that are approved for use in Florida's public waters and are labeled for use with hydrilla and/or were used in 2013 for hydrilla control. Check with the UF/IFAS Pesticide Information Office for label changes.

Active ingredient common name	Examples of trade names	Mode of action	Maximum use rate for submersed treatments ¹	Used in Florida for hydrilla control in 2013 ²	Amount of active ingredient applied for hydrilla control in Florida in 2013 (lbs) ^{2,3}
Bispyribac-sodium	Tradewind®	Systemic	0.05 mg/L	Yes	347
Copper	Komeen®	Contact	1.00 mg/L	No	n/a
Diquat	Reward® Tribune™	Contact	0.37 mg/L	Yes	40,192
Endothall	Aquathol® Hydrothol®	Contact	5.00 mg/L	Yes	220,903
Flumioxazin	Clipper™	Contact	0.40 mg/L	Yes	1,079
Fluridone	Sonar®	Systemic	0.15 mg/L	Yes	251
Imazamox	Clearcast®	Systemic	0.50 mg/L	No	n/a
Penoxsulam	Galleon®	Systemic	0.15 mg/L	Yes	764

¹ Information extracted from the UF/IFAS Center for Aquatic and Invasive Plants website. URL: <http://plants.ifas.ufl.edu/manage/control-methods/chemical-control/selective-application-of-aquatic-herbicides>

² Information extracted from the Florida Fish and Wildlife Conservation Commission Annual Report of Pollutant Discharges to the Surface Waters of the State from the Application of Pesticides, 2013

³ n/a = not applicable

The use of trade names in this publication is solely for the purpose of providing specific information. UF/IFAS does not guarantee or warranty the products named, and references to them in this publication do not signify our approval to the exclusion of other products of suitable composition. All chemicals should be used in accordance with directions on the manufacturer's label.



To become familiar with laws, regulations, and herbicide safety, you can visit the UF/IFAS Pesticide Information Office online.

URL: <http://pested.ifas.ufl.edu>
(Scan the QR code to connect to the website.)

BISPYRIBAC-SODIUM

Bispyribac-sodium is a systemic herbicide that accumulates in the growing regions of the plant and inhibits enzymes that are necessary for amino acid production and plant growth. Bispyribac-sodium is not fast acting. The plant stops growing and eventually dies over a period of several months.

Advantages and Disadvantages of the Aquatic Herbicide Bispyribac-sodium

Advantages: low non-target toxicity, fairly selective for submersed weeds, reduced impact on other organisms due to slow plant death and decay

Disadvantages: slow acting

COPPER

Copper is used mainly for algae control, but there are several products labeled for use with hydrilla. It is a contact herbicide that kills a range of aquatic plants and algae. Copper is fast acting. Although it is an essential nutrient for plants, an abundance of copper interferes with plant metabolism.

Chelated copper products are more effective as they stay in solution longer than salts that were traditionally used. The use of copper is permitted only in waterways with no alternative, as it is toxic at low doses to fish and mollusks. However, copper is often used in drinking-water sources where use of other herbicides is restricted.

Advantages and Disadvantages of the Aquatic Herbicide Copper

Advantages: fast acting, can be used in drinking-water sources

Disadvantages: toxic to fish and mollusks, broad-spectrum herbicide, quick kill impacts other organisms due to plant death and decay, will accumulate in sediment

DIQUAT

For submersed plants like hydrilla, diquat acts as a contact herbicide. Diquat is fast acting and kills by producing free radicals that interfere with photosynthesis. Diquat does not work well in muddy waters as the positively charged chemical is quickly absorbed by negatively charged peat and clay.

There are strict restrictions on the use of diquat around drinking water sources. For example, in flowing water, diquat cannot be applied 1,600 ft upstream or 400 ft downstream of a potable water intake site. For hydrilla control, diquat needs to be used in combination with an herbicide with another mode of action, for example, copper or endothall.

Advantages and Disadvantages of the Aquatic Herbicide Diquat

Advantages: fast acting

Disadvantages: restrictions around drinking water sources, cannot be used alone for hydrilla control, does not work well in muddy water, quick kill impacts other organisms due to plant death and decay

ENDOTHALL

Endothall is a fast-acting contact herbicide that is used primarily for submersed aquatic vegetation. It is absorbed quickly by the leaves that come in contact with the chemical. Endothall has several modes of action—it can inhibit plant protein and lipid synthesis, disrupt cell membrane integrity, and reduce the activity of certain plant enzymes. Depending on the concentration, endothall usually kills the plant within 12-36 hours.

Control is relatively long, with treatments usually required no more than every six months. There are two types of endothall, potassium endothall and amine endothall. Although amine endothall is slightly more effective on plants, it is 200-400 times more toxic to fish and so should be used with caution. Due to the development of resistance to other chemicals in hydrilla, endothall is currently being used extensively in Florida.

Advantages and Disadvantages of the Aquatic Herbicide Endothall

Advantages: fast acting, long duration of control

Disadvantages: quick kill impacts other organisms due to plant death and decay

FLUMIOXAZIN

Flumioxazin is a contact herbicide that inhibits photosynthesis causing cell death. Exposed plant tissue becomes yellow and brown. Following four hours of exposure, treated plants will die within a few days to a week.

Advantages and Disadvantages of the Aquatic Herbicide Flumioxazin

Advantages: fast acting

Disadvantages: quick kill impacts other organisms due to plant death and decay

Herbicide — a substance that kills weeds (usually a chemical compound)

Resistance [n.], resistant [adj.] (to herbicides) — the ability of a plant to survive the exposure to a typically lethal dose of herbicide

Submersed — a plant with most leaves growing underwater; flowers and some of the leaves may float on the water surface



Herbicide — a substance that kills weeds (usually a chemical compound)

Resistance [n.], resistant [adj.] (to herbicides) — the ability of a plant to survive the exposure to a typically lethal dose of herbicide

FLURIDONE

Fluridone is a systemic herbicide that enters the plant through the roots and shoots and causes the destruction of chlorophyll preventing photosynthesis. The shoot tips of treated plants become pink or white. Fluridone is not fast acting, and the plant eventually dies from starvation.

For optimum control of hydrilla, the concentration must be maintained for several months. Following extensive use of fluridone for hydrilla management, fluridone-resistant hydrilla is now present in Florida. It is advisable to perform a pre-application bioassay to determine the resistance of local hydrilla populations to fluridone prior to large-scale applications.

Advantages and Disadvantages of the Aquatic Herbicide Fluridone

Advantages: low non-target toxicity, area controlled is larger than area treated, long-term control, reduced impact on other organisms due to plant death and decay

Disadvantages: slow acting, fluridone resistance, broad spectrum

IMAZAMOX

Imazamox is a systemic herbicide with a similar mode of action as bispyribac-sodium. The herbicide penetrates plant tissue and inhibits amino acid production in growing regions. Like bispyribac-sodium, imazamox is not fast acting. Although growth is affected relatively quickly and the plant may yellow after 1-2 weeks, death will not occur for several weeks. Imazamox is used at high doses to kill hydrilla and at lower doses as a growth regulator.

Advantages and Disadvantages of the Aquatic Herbicide Imazamox

Advantages: low non-target toxicity, reduced impact on other organisms due to slow plant death and decay, regulates growth immediately and at low doses

Disadvantages: slow acting

PENOXsulAM

Penoxsulam is a systemic herbicide with the same mode of action as bispyribac-sodium and imazamox in that it prevents growth by inhibition of amino acid production. Plant death occurs very slowly, and herbicide concentration must be maintained for 3-4 months to achieve control. Combination with herbicides with other modes of action, such as endothal, may reduce this exposure time and provide faster results.

Advantages and Disadvantages of the Aquatic Herbicide Penoxsulam

Advantages: low non-target toxicity, reduced impact on other organisms due slow to plant death and decay

Disadvantages: slow acting, better in combination

Pesticide — a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals

Understanding Herbicide Labels

All containers of herbicide must have a label that provides the applicator with specific information that they need to apply the product safely, legally and effectively. See Figure 36 for an example of an aquatic herbicide label.

It is against the law to change, remove or destroy a label. It is also against the law to misuse an herbicide by not following the specifications on the label. **The herbicide is only approved to be used as stated on the label. It is not advice. It is the law!** Using the herbicide in any other way is a violation of federal and state law and applicators that misuse herbicides could face imprisonment.

All labels should provide:

- Product information
- Safety information
- Environmental information
- Directions for use
- Storage and disposal instructions

PRODUCT INFORMATION

The product information is usually the first part of the label (Figure 36). The first item to consider is the **EPA use classification**. Herbicides can be classified as restricted use or general use. Restricted use pesticides (also called RUPs) will have a notification at the top of the first page of the label. The label in Figure 36 is not restricted use and so does not list the notification.

Restricted use herbicides may only be purchased and applied by a pesticide applicator who is certified and licensed in the state of Florida. To our knowledge, there are no restricted use pesticides currently registered for use as aquatic herbicides in Florida. General use herbicides may be purchased by the public and do not necessarily require certification. However, if you are applying herbicides for your work, your employer may require that you become certified and licensed.

The **brand or trade name** is the next item to notice. This is the name given to the product by the manufacturer. The brand name often has an abbreviation to indicate the type of formulation. For example, G usually means granular, D for dusts, WP for wettable powder, and E or EC for emulsifiable concentrate. The label that you are reading is specific to that product and formulation and cannot be used to apply another product even if the active ingredient is the same.

Herbicide — a substance that kills weeds (usually a chemical compound)

Pesticide — a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals

Underneath the brand name you will usually find the **active ingredient** list, which includes active and other ingredients with a percentage by weight amount. The active ingredients are the part of the product that is having the effect on the target weed. The ingredients may be provided as common names (e.g., imazamox) or chemical names (e.g., 2-[4,5-dihydro-4-methyl-4-(1-methylethyl)-5-oxo-1H-imidazol-2-yl]-5-(methoxymethyl)-3-pyridinecarboxylic acid).

There should be two pieces of **EPA information**, the EPA registration number and the EPA establishment number. The EPA registration number indicates that the product has met federal registration requirements. The number provides information about the product, manufacturer and distributor. The EPA establishment number identifies the facility that formulated the product.

The manufacturer will provide their **name and address** and a **warranty disclaimer**. The warranty disclaimer usually states that the product conforms to the chemical description on the label and is fit for purpose. The warranty does not extend to misuse of the product or use under abnormal weather conditions.

SAFETY INFORMATION

The safety information provided on an herbicide label is based on extensive chemical and biological testing and should be carefully considered. There will usually be a **Child Hazard Warning** as children are the most at risk of pesticide poisoning (see Figure 36).

There will be a **signal word**, this indicates the toxicity of the product (not the active ingredient) to humans. The signal word may be: DANGER POISON, DANGER, WARNING or CAUTION. DANGER POISON indicates that the product is highly toxic and may cause illness through exposure to the skin, ingestion or inhalation. Ingestion of a few drops to a teaspoonful of a product labeled with DANGER POISON (Category I) can be lethal. Products labeled DANGER may irritate the eyes and skin if these areas are exposed.

Products labeled WARNING (Category II) are moderately toxic and ingestion of one teaspoon to one ounce can be lethal. Products labeled CAUTION may be category III or IV. Category III is slightly toxic, ingestion of one ounce to one pint can be lethal. Category IV is relatively non-toxic and ingestion of one pint to one pound can be lethal. Any category I products labeled DANGER, must also have a **statement of practical treatment** that describes what to do in an emergency should exposure occur. Labels of less toxic products may also provide first aid instructions.

If an exposure occurs and the label advises you to seek medical attention you should take the pesticide label with you to the hospital. If this is not possible make sure that you have the brand name and manufacturer so that the medical professional can look up the information.

The **potential hazards to humans and animals** will be provided, the specific hazard, route of exposure and precautions to avoid exposure will be given. For example, “Harmful if swallowed or inhaled. Avoid breathing dusts.” The **physical or chemical hazards** will be listed somewhere on the label, such as risk of fire or explosion. The label will provide precautionary measures to reduce the risk of the hazard. For example, if a product is flammable the label may read “do not use or store near heat or open flames.”

In addition to these precautions, information about suggested **personal protective equipment (PPE)** also will be provided. Examples of PPE include specific types of clothing, eye wear, waterproof gloves and chemical resistant shoes. The list provided on the label is the **minimum** that you should use when applying the herbicide.

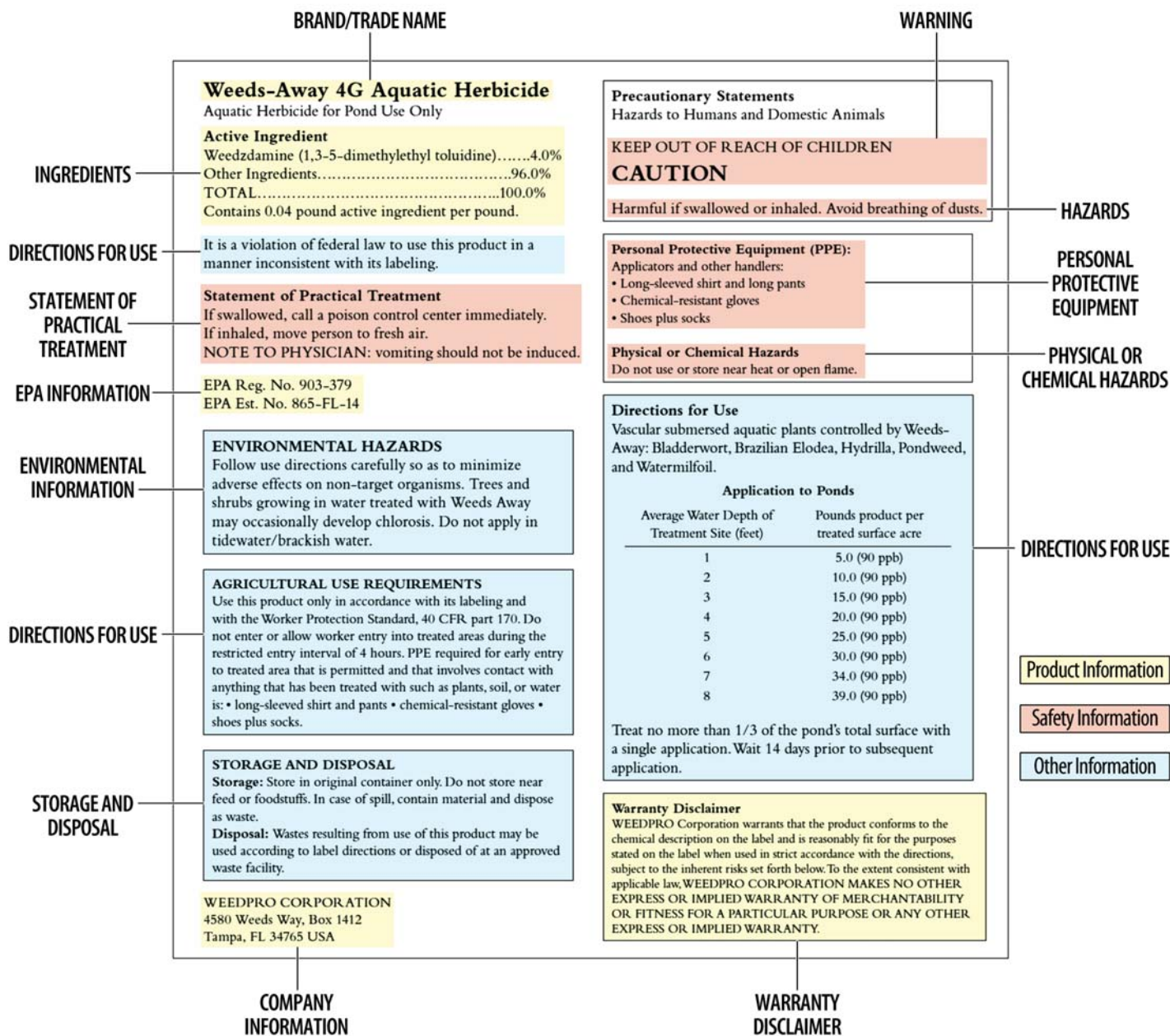


Figure 36. Example herbicide label with important product, safety, and environmental information sections highlighted as well as directions for use, storage, and disposal. Label used with permission from the UF/IFAS Center of Aquatic Invasive Plants.

ENVIRONMENTAL INFORMATION

The section of the label on **environmental hazards** will detail any potential hazards for other plants, animals or the environment (see Figure 36). Information will be provided about the hazards as well as ways to avoid impacts on non-target organisms. If the product has been shown to be toxic to honeybees or fish this will be mentioned in this section.

DIRECTIONS FOR USE

There should be a statement emphasizing to the user the legal importance of following the label instructions (see Figure 36). Products that will be used in agriculture should have a statement that informs the user that Worker Protection Standard 40 CFR Part 170 applies. This section should also provide specific information to the worker about re-entry times, training required, emergency assistance and required PPE.

When considering the rest of the directions for use section, the user should look for several pieces of information:

1. Where the product can be applied
2. The amount of product to apply
3. How the product should be applied
4. The timing and frequency of applications
5. Limitations on water use following treatment, e.g. drinking, swimming, fishing, irrigation and livestock watering
6. The target pests
7. Any other information specific to the product

Aquatic herbicide applicators always check item number 1 first to find out whether the product is labeled for the site or not. If it is not, there is no reason to read any further, the product may not be used.

The use of a product to target a pest that is not included in the list on the label could result in an off-label application and misuse of the product. The product may not work, and the user assumes all risks associated with the application. However, according to the 2013 Florida Statutes, title XXXII, chapter 487, 487.031, it is not unlawful to “apply a pesticide against any target pest not specified in the labeling if the application is to a crop, animal, or site specified on the label or labeling, provided that the label or labeling does not specifically prohibit the use on pests other than those listed on the label or labeling.”

STORAGE AND DISPOSAL INSTRUCTIONS

The instructions will be given for correct **storage** of the product (see Figure 36). Pay particular attention to recommended temperatures. Storing the product above or below this range may result in the active ingredient becoming ineffective. Some herbicides should not get wet, particularly dusts or granular formulations, in this case instructions will be provided to store the product in a dry place.

Information about proper **disposal** of leftover product or product containers should be provided. Usually any unaltered leftover product that is not used according to the label should be returned to the manufacturer. Product that has been altered, such as diluting or mixing with a carrier for spraying, should be disposed of in an approved waste disposal facility. Some empty containers may be returned to the manufacturer. Otherwise the user may be advised to triple rinse containers and discard them into sanitary landfill. Remember—for every product the suitable methods for disposal will be different and even for the same product they may change over time. Always check the label before disposing of any product or product containers.

Aquatic Herbicide Applicator License

All people that apply or supervise the application of restricted use pesticides in Florida must be certified and licensed by the Florida Department of Agriculture and Consumer Services (FDACS). However, it is recommended that all applicators who use herbicides for management of aquatic plants should be licensed. The FDACS has a special licensing category called Aquatic Pest Control for this purpose. Most people that are employees of companies that are contracted to perform application of herbicides for aquatic pest control, i.e., application to standing or running water, banks or shorelines, are required to be certified.

There are two classes of license including a public applicator, which is an applicator employed by public or government agency and a commercial applicator, which is an applicator that is licensed to apply herbicides on any property. The certification requirements are the same for both, but the limits of use and fees are different. Both classes must pass two examinations, a general knowledge about pesticides use exam and the Aquatic Pest Control specific exam.

These exams can be taken at UF/IFAS County Extension offices by appointment. Call to make an appointment and check that your local Extension office can perform both examinations. The study materials can be purchased from the UF/IFAS Extension Bookstore.

Once you have passed the two exams, you will receive notification from the FDACS Certification and Licensing Office and your license will be valid for four years.

In order to renew your license, without needing to retake the exam, you must complete Continuing Education Units (CEUs). To renew your license you need four core CEUs plus category CEUs depending on the category of applicator. Aquatic Pest Control applicators need 16 category CEUs in addition to the four core CEUs.

Continued Education Units are earned by attending in person or online approved CEU classes. Education providers such as County Agricultural Extension Offices offer training programs with CEU credits. Applicators must keep track of CEUs earned and submit documentation when they renew their license (CEU Record of Attendance forms).

An excellent option for applicators to gain CEUs is the Hydrilla IPM CEU approved course. Applicators in the Aquatic Pest Control, Private Applicator Agricultural Pest Control, and Right-of-way Pest Control can earn one category CEU by completion of our online training. Applicators can download the workbook and complete the questions while watching the online lessons. The Hydrilla IPM workbook and training enrollment form then needs to be mailed in and a signed completed CEU Record of Attendance form will be returned.



Visit the UF/IFAS Hydrilla CEU course webpage to download the workbook, watch the lessons, and find more information about the course. URL: <http://pesticide.ifas.ufl.edu/courses/HydrillaIPM.shtml>
(Scan the QR code to connect to the webpage.)

Herbicide — a substance that kills weeds (usually a chemical compound)

Pesticide — a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals

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Pesticide — a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals



For the general pesticide use application exam, you should study *Applying Pesticides Correctly: A Guide for Pesticide Applicators (CORE)*. URL: <http://ifasbooks.ifas.ufl.edu/p-104-applying-pesticides-correctly-a-guide-for-pesticide-applicators-core.aspx>
(Scan the QR code to find the guide online.)



For the Aquatic Pest Control exam, you should study the *Aquatic Pest Control Training Manual (Aquatic Category Exam)*. URL: <http://ifasbooks.ifas.ufl.edu/p-106-aquatic-pest-control-training-manual-aquatic-category-exam.aspx>
(Scan the QR code to find the manual online.)



Applicators can search for classes using the CEU Class Search. URL: <http://app1.flaes.org/ceu/AvailableClassSearch.asp>
(Scan the QR code to connect to the search engine.)



Find more information via the UF/IFAS Extension Bookstore. URL: <http://ifasbooks.ifas.ufl.edu>
(Scan the QR code to connect to the website.)

Summary Table: Advantages and Disadvantages of Herbicides Available for Hydrilla Control

Always be sure that the application that you are attempting is legal and has been approved by local regulatory agencies. The table below summarizes different features of the herbicides that are available for hydrilla control in Florida.

Herbicide	Action	Mode of action	Speed of action	Host specificity ¹	Non-target effects on animals	Restrictions			
						Potable water	Recreation ²	Irrigation	Livestock watering
Bispyribac-sodium	Systemic	Inhibits amino acid production	Slow	Low	Minimal	No	No	Yes, 1 ppb	Yes, 1 ppb
Copper	Contact	Interferes with metabolism	Fast	Low	Yes, aquatic invertebrates and fish	No	No	No	No
Diquat	Contact	Inhibits photosynthesis	Fast	Low	Yes, aquatic invertebrates	Yes, 1-3 days	No	Yes, min. 5 days	Yes, 1 day
Endothall	Contact	Several, interferes with metabolism	Fast	Low	No ^{3,4}	Yes, do not apply within 600 ft	No	Yes, min. 7 days	Depends on formulation
Flumioxazin	Contact	Inhibits photosynthesis	Fast	Low	Yes, aquatic invertebrates and fish	No	No	Yes, min. 5 days	No
Fluridone	Systemic	Inhibits photosynthesis	Slow	Low	Yes, aquatic invertebrates	Yes, if more than 20 ppb	No	Yes, 7-30 days	No
Imazamox	Systemic	Inhibits amino acid production	Slow	Low	No ^{4,5}	Yes, if more than 50 ppb	No	Yes, 24 hours	No
Penoxsulam	Systemic	Inhibits amino acid production	Slow	Low	No ⁴	No	No	Yes, 1 ppb	No

¹ Host specificity of the chemical, without considering timing and dose

² Recreation, e.g. swimming and fishing

³ When the less toxic potassium endothall is used, amine endothall is 200-400 times more toxic

⁴ Non-toxic to non-target animal species when applied at the recommended label rate and following application guidelines

⁵ Toxic to bees so avoid drift during application

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Chapter 5



Proposals for Integrated Hydrilla Management

By Emma N.I. Weeks and
Verena-U. Lietze

Subsection by Amy L. Giannotti
and Marissa L. Williams

CONTENTS

Combining Different Tactics	66
Resistance Management	69
Management in Different Water Body Types	72

Resistance [n.], resistant [adj.] (to herbicides) — the ability of a plant to survive the exposure to a typically lethal dose of herbicide



Visit the UF/IFAS Hydrilla IPM RAMP website for our most up-to-date recommendations. URL: <http://entomology.ifas.ufl.edu/hydrilla> (Scan the QR code to connect to the website.)

This chapter presents current research that explores the options of combining—that is, integrating—different control methods in comprehensive hydrilla integrated pest management (IPM) plans. Furthermore, it addresses the problem of resistance and introduces methods of hydrilla management in different types of water bodies.

Combining Different Tactics

The next three sections will offer you some insight into the research that is involved in developing an IPM plan. Please also refer to the IPM section starting on page 4. IPM will help you combat hydrilla in new ways. We are hopeful that the tactics and approaches described in this guide will help you find reduced-risk methods for the management of hydrilla, so you can address potential concerns from stakeholders like those we have listed on page 131.

Hydrilla Tip Mining Midge and a Plant Pathogenic Fungus

Results from short-term aquarium experiments showed that the hydrilla tip mining midge, *Cricotopus lebetis*, is compatible with the plant pathogenic fungus *Mycoleptodiscus terrestris* (Mt). Moreover, within only 28 days, a synergistic effect was seen (Figure 37). The combined treatment with a high dose of Mt fungus and a low density of hydrilla tip mining midge larvae significantly reduced hydrilla biomass by almost 80% when compared with the untreated control.

This approach is a combination between two different biological control agents. Our hypothesis is that the midge larvae feeding in the hydrilla tips open up the plant tissue for increased penetration of the fungus. It is important to note that careful selection of dosages seems necessary to achieve this effect. You can get more information on the hydrilla tip mining midge on page 45 and on the plant pathogenic Mt fungus on page 47.

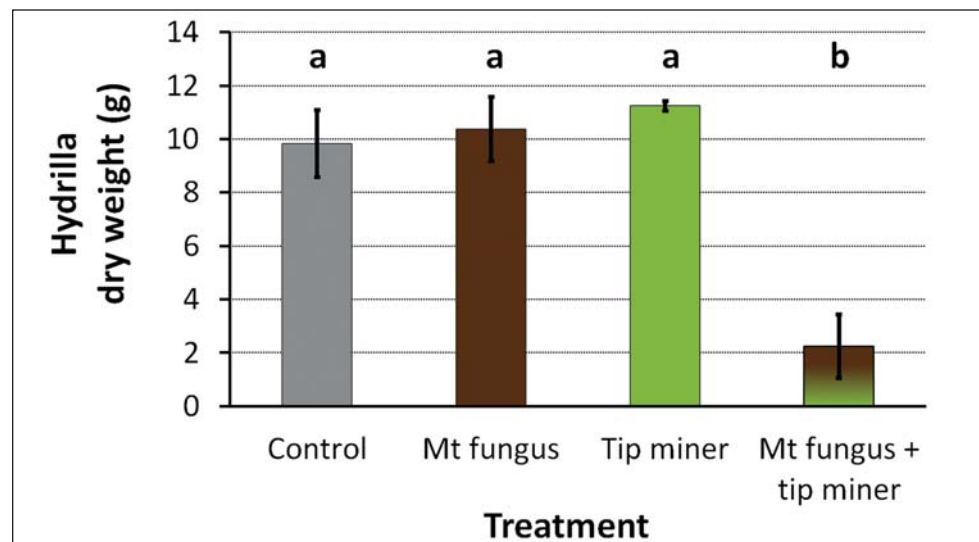


Figure 37. The graph shows the biomass (dry weight) of hydrilla 28 days after application of the plant pathogenic fungus *Mycoleptodiscus terrestris* (Mt) and/or the hydrilla tip mining midge in 55-Liter aquariums. Controls received no treatment. Different letters indicate statistically significant differences when compared with the control (ANOVA and Fisher's LSD test, $\alpha < 0.05$). The experiments were conducted at the U.S. Army Engineer Research and Development Center (ERDC) in Vicksburg, Mississippi. Special thanks to Judy Shearer (ERDC) and James Cuda (UF/IFAS) for sharing their results.

Hydrilla Tip Mining Midge and the Herbicide Imazamox

Results from short-term aquarium experiments showed that the hydrilla tip mining midge, *Cricotopus lebetis*, is compatible with the herbicide imazamox. Furthermore, within only 28 days, the combined treatment had a synergistic effect on hydrilla (Figure 38). It significantly reduced the numbers of hydrilla shoot tips by about 50% when compared with the untreated control. Individual treatments, however, were not effective.

This approach is a combination between biological and chemical control. Our hypothesis is that the imazamox treatment causes the branching of the hydrilla, which provides additional feeding sites for the midge larvae. The midge larvae then feed in the hydrilla shoot tips causing them to die and break off. These results demonstrate the importance and effectiveness of integrated approaches to weed control. You can get more information on the hydrilla tip mining midge on page 45 and on the herbicide imazamox on page 56.

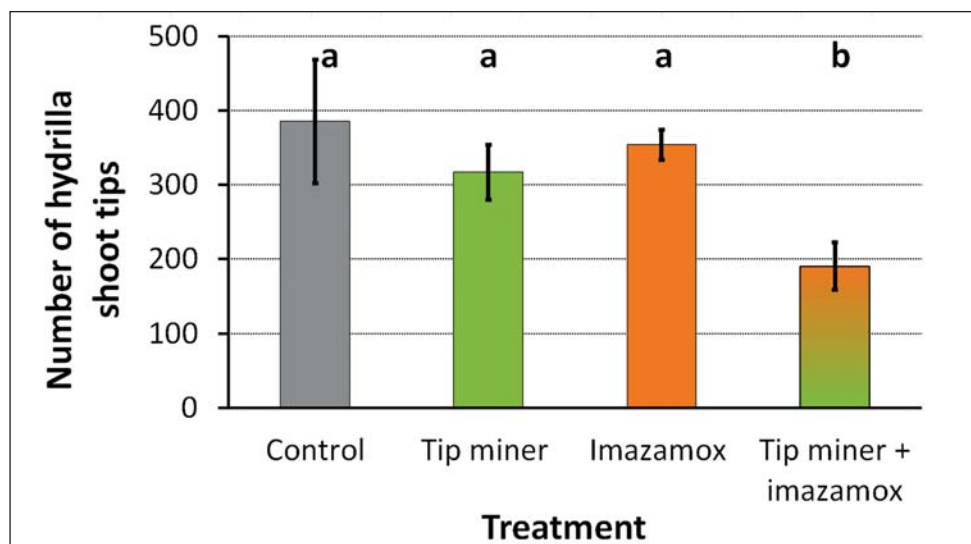


Figure 38. The graph shows the number of hydrilla shoot tips counted 28 days after application of the hydrilla tip mining midge and/or imazamox in 55-Liter aquariums. Controls received no treatment. Different letters indicate statistically significant differences when compared with the control (ANOVA and Fisher's LSD test, $\alpha < 0.05$). The experiments were conducted at the U.S. Army Engineer Research and Development Center (ERDC) in Vicksburg, Mississippi. Special thanks to Judy Shearer (ERDC) and James Cuda (UF/IFAS) for sharing their results.

Plant Pathogenic Fungus and Herbicides

Compatibility of the plant pathogenic fungus *Mycoleptodiscus terrestris* (Mt) with aquatic herbicides has been researched extensively since the 1990s and was overseen by the U.S. Army Corps of Engineers at the Engineer Research and Development Center in Vicksburg, Mississippi.

Results from laboratory experiments showed that the Mt fungus is compatible with several herbicides including diquat, endothal, and fluridone. Within only 21 to 35 days, a synergistic effect was seen. For example, combined treatment of hydrilla with the Mt fungus and fluridone significantly reduced hydrilla biomass by 92% when compared with the untreated control and by over 80% when compared with the individual treatments (Figure 39).

Herbicide — a substance that kills weeds (usually a chemical compound)

Synergistic effect — the effect caused by two methods or agents together is greater than the sum of the effects of each individual method or agent

This approach is a combination between biological and chemical control. Our hypothesis is that the combination of the fungus with herbicides provides additional control because the modes of action are different. Any tolerance or resistance of hydrilla to either tool used alone could result in treatment failure. When the tools are combined, the plant has less chance to survive. When the fungus is applied alone, it kills only the part of the plant that it contacts. When the herbicide is applied alone, there may be issues with tolerance or resistance.

In the field, these tools are highly complementary when applied in combination, particularly with slow-acting synergistic herbicides. The Mt fungus provides relatively fast initial control, and the herbicide provides a long-lasting effect on the reduced biomass that remains. You can get more information on the Mt fungus on page 47 and on the herbicides diquat, endothall, and fluridone on pages 54-56.

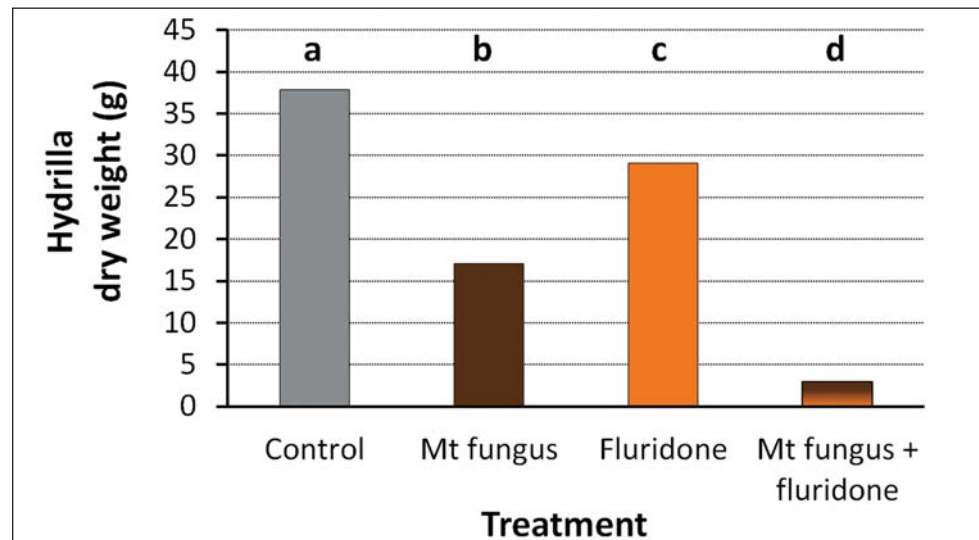


Figure 39. The graph shows the biomass (dry weight) of hydrilla 35 days after application of the plant pathogenic fungus *Mycocleptodiscus terrestris* (Mt) and/or fluridone in 55-Liter aquariums. Controls received no treatment. Different letters indicate statistically significant differences when compared with the control (ANOVA and Fisher's LSD test, $\alpha < 0.05$). The experiments were conducted at the U.S. Army Engineer Research and Development Center (ERDC) in Vicksburg, Mississippi. Special thanks to Judy Shearer (ERDC) for sharing her results.

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Resistance Management

Resistance management has become a major issue in hydrilla control. For many years, researchers and applicators did not worry about the development of herbicide resistance in dioecious hydrilla populations because they assumed that only one biotype resided in the U.S. This biotype demonstrated high susceptibility to available herbicides. However, time has proven these assumptions wrong as more and more populations of resistant biotypes of hydrilla emerge.

What Is Resistance?

Within a given population, a plant species that once was susceptible to a given rate of a given herbicide may no longer be controlled by exposure to this herbicide. Such **resistance** will occur after repeated exposure to the herbicide, because only those individuals or biotypes with a trait that helps them survive the exposure to the herbicide (often due to a slight genetic difference) will continue to grow and reproduce. As a consequence, application rates have to be increased, and eventually, the use of the herbicide is no longer feasible. The entire population is now resistant to this herbicide.

A plant species that is resistant to a certain herbicide may also show **cross-resistance**, that is, resistance to another herbicide with a similar mode of action. For example, hydrilla populations that are resistant to fluridone also are resistant to norflurazon, because both herbicides have the same mode of action—they inhibit the enzyme phytoene desaturase.

Resistance is not the same as **tolerance**. A plant species that shows herbicide tolerance has never been susceptible to this herbicide. Such tolerance can be based on genetically determined physical or biochemical traits (such as a thick cuticle or a detoxifying metabolic pathway) that protect the plant from the effects of the herbicide.

Herbicide Resistance in Hydrilla Populations

Various chemical, mechanical, and biological methods have been investigated for managing hydrilla infestations in an attempt to control the explosive growth of the weed, but none to date (2014) have been as effective as the synthetic chemical herbicide fluridone (trade name Sonar®).

About a decade ago, it was discovered that hydrilla had developed resistance to fluridone, which is a systemic herbicide for aquatic systems approved by the U.S. Environmental Protection Agency for managing this submersed invasive aquatic weed. In 2011, the first reports documented resistance of hydrilla to endothall, a fast-acting contact herbicide also approved for aquatic use in the U.S. The resistance problem is increasing, and new tools and tactics to cope with this problem need to be developed.

The spread of resistant hydrilla biotypes to other water bodies within the U.S. is inevitable. What is not known is how to effectively control hydrilla as it loses its susceptibility to fluridone and endothall. Lack of such knowledge is a critical problem because, until it becomes available, the spread of resistant hydrilla biotypes and the higher herbicide concentrations needed to control them will increase the cost to state and federal programs to manage this weed. In addition, the higher herbicide rates will pose a risk to non-target organisms including agricultural and ornamental plants that depend on safe water for irrigation.

Biotype — a form of the same plant species that shows special characters (for example, presence/absence of male or female flowers, resistance to a chemical herbicide, tolerance to extreme temperatures)

Dioecious — female and male flowers occur on different plants

Herbicide — a substance that kills weeds (usually a chemical compound)

Resistance [n.], resistant [adj.] (to herbicides) — the ability of a plant to survive the exposure to a typically lethal dose of herbicide

Submersed — a plant with most leaves growing underwater; flowers and some of the leaves may float on the water surface



The Importance of Product Rotation

Reliance on one product is no longer acceptable in aquatic plant control. Given the costs, labor, and time involved in developing novel herbicides, we need to make sure that products, once they are approved for use, yield the desired control results. Rotating applications between herbicides of different modes of action will support this objective.

We hope you will go beyond thinking about herbicide products as your sole management option, remember the IPM continuum (see page 4). We cannot stress enough the importance of developing an IPM plan. You should have several different tactics at work in your management area to reduce the impact of hydrilla.

Furthermore, when you notice that hydrilla populations in your area are no longer susceptible to previously effective herbicide treatments, immediately contact your local aquatic weed specialist (refer to the Contacts for Plant Identification and Management Advice section on page 126).

A Case Report on Integrated and Resistance Management of Hydrilla in Florida

By Amy L. Giannotti and Marissa L. Williams

The information in this section is not based on replicated research performed by the University of Florida. This information was provided to share field techniques that are being applied by our peers (Figure 40) and are showing promising results.

We have been actively involved in hydrilla management here in Winter Park since the late 1960s. Our 23-year-old stormwater utility program fully funds hydrilla management and water quality related issues on 23 lakes within the City. Historically, hydrilla management was accomplished via the use of mechanical harvesters in the 1960s, and then as it became increasingly difficult and expensive to control, the City incorporated the use of endothall products that were used regularly for a number of years in spot-treatment scenarios.

When fluridone was introduced in the 1990s, Winter Park began using this systemic herbicide intermittently with endothall to rotate the mode of action that was impacting the plants. After two failed fluridone treatments in 2005 and 2007, fluridone was discontinued. The Winter Park Chain of Lakes entered the FWC funded program in 2008, and in order to combat the rapidly spreading hydrilla, we stocked the lakes with a relatively low rate of Asian grass carp in conjunction with continued herbicide treatments.

In 2009 and 2010, several whole-lake treatments were done using endothall at low rates, and monitoring was conducted before, during, and after the treatments to ensure target concentrations were achieved and to provide further data on the use of endothall in cold weather whole-lake scenarios to Dr. Netherland at the U.S. Army Corps of Engineers. Ironically, both of these treatments yielded unexpected results where hydrilla was still growing and showed no sign of impact even though lethal doses were reached in the water column. Evaluation of these plant communities confirmed that these two lakes harbored



Figure 40. Photo of the authors Marissa Williams (left) and Amy Giannotti (right) in action at a workshop in Winter Park, Florida. Photograph by Tim Egan, City of Winter Park.



the only known population of hydrilla which is not only resistant to fluridone, but is also resistant to endothall.

Since then, our reliance on Asian grass carp has increased, and we are utilizing new herbicides on the market (Clipper™ containing flumioxazin, Clearcast® containing imazamox) as well as unique combinations of these new and old herbicides, and we will be incorporating some of the new herbicide products (Tradewind® containing bispyribac-sodium, Galleon® containing penoxsulam, and Clearcast® containing imazamox) in the fall of 2014 should the need arise. However, unlike the contact herbicides, which are applied to localized areas of the lake and have minimal irrigation restrictions, these new products treat the hydrilla in the entire lake all at once. As such, the irrigation restrictions associated with the use of these new herbicides are lengthy. Based on current label requirements, it may be necessary to cease irrigation for up to four months.

Few people realize that there are small clusters of naturally resistant plants living within a given population of hydrilla. Repeated use of the same products over time kills off only those plants that are susceptible, leaving behind the resistant plants to regenerate and eventually dominate the community. By altering the modes of action that chemically affect the plant, we hope that these uniquely tolerant populations will be impacted by the new herbicides and combinations, thus reducing the likelihood that multiple resistance issues will develop again in this system.

How Can You Help Prevent Herbicide Resistance?

Here are some important actions for you to keep in mind:

- Use an integrated management plan that includes tactics other than chemical control (see the Developing an IPM Plan for Aquatic Weed Infestations section on page 5).
- Always consider non-chemical methods first (such as physical or mechanical control and biological control agents).
- If you have to use chemical control methods (i.e., herbicides), rotate products that are suitable for hydrilla control (see Summary Tables on pages 53 and 63 for more details).
- Use herbicides only when water and weather conditions are suitable and according to the instructions on the label.
- Apply herbicides at the recommended rates, so that plants are not exposed to ineffective concentrations of the active ingredient.

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Herbicide — a substance that kills weeds (usually a chemical compound)

Resistance [n.], resistant [adj.] (to herbicides) — the ability of a plant to survive the exposure to a typically lethal dose of herbicide



Distribution — the geographical range in which the plant occurs

Herbicide — a substance that kills weeds (usually a chemical compound)



An overview of Florida’s waters is provided by the UF/IFAS Center for Aquatic and Invasive Plants. URL: <http://plants.ifas.ufl.edu/manage/overview-of-florida-waters/introduction> (Scan the QR code to connect to the website.)

Management of Hydrilla in Different Water Body Types

QUOTE: “[I’d like to see] information on what works best to control hydrilla in moving/quiescent water.” —G.A.

This quote is an important request because the extent of water flow will affect the distribution and concentration of herbicides and biological control organisms. It also impacts the direction in which hydrilla may spread. Different water bodies therefore require different approaches to hydrilla management.

Characteristics of Different Water Bodies

The following sections describe key characteristics of different water bodies and how they may affect aquatic weed management. More detailed information is available through the UF/IFAS Center for Aquatic and Invasive Plants. Scan the QR code on the left to connect directly to the website.

Rivers and Streams

Florida’s landscape includes almost 1,700 rivers and streams. They are bodies of moving freshwater originating from springs and ranging in width from a few feet to about 2 miles. They are essential for the transport of nutrients and sediment to the abundant wetland areas of our state. Besides their many ecological functions, rivers support a number of commercial, agricultural, and recreational uses.

Canals

Canals span thousands of miles across Florida. They are artificially constructed waterways that can be as small as a few feet wide and deep and as large as several hundred feet wide and up to 35 feet deep. Canals support various functions, such as navigation between natural water bodies, irrigation, flood and drainage control, and recreation.

Lakes and Ponds

Florida harbors almost 8,000 lakes and ponds. Each one is a unique ecosystem with a characteristic shape, hydrology, geology, flora, and fauna. A pond is defined as a body of water that is surrounded by land and shallow enough so that an adult could wade through the water. In general, a lake is defined as a large (at least 4 acres) body of water that is surrounded by land and is deeper than a pond—so deep that an adult could not wade through the water.

Some lakes and ponds have formed naturally, others were constructed artificially. Lakes and ponds have important functions including irrigation, flood control, drinking water supply, recreation, and navigation. Depending on the inflow and outflow of water and on wind and other factors, some lakes and ponds, although seemingly still on the surface, have considerable water movement. However, the movement is generally much less than that observed in rivers and streams.

Impoundments

An impoundment is a body of water that is confined within an enclosure. It is formed artificially “by the construction or excavation of a basin or the obstruction of stream flow in such a manner as to cause the collection of a body of water which would not have formed under natural conditions” (North Carolina Administrative Code 1990).

Impoundments serve as reservoirs of water that can be used for irrigation or hydraulic processes. Large impoundments generally exhibit considerable water movement because of thermal currents and/or wind.



Management of Submersed Weeds in Different Waters

The following sections provide an overview of management tactics that could be combined in different types of water bodies.

Management of Submersed Weeds in Flowing Water

Aquatic vegetation is less of a problem in rivers and streams than in other water bodies such as lakes and ponds. However, when excessive plant growth occurs or invasive weeds do grow in these areas, there can be a big effect on water flow and use of the waterway can be restricted (Figure 41). The movement of water from one area to another poses several problems in aquatic weed control. In particular, although submerged weeds are rooted in the sediment and not as affected by the movement as floating weeds, surface mats like those produced by hydrilla may break off and float to new areas, where they can start new infestations.

Although **physical control** methods are difficult to implement in flowing water, they are often applied. *Hand removal* and *suction harvesting* are likely to be the most appropriate methods to use in these sensitive ecosystems. In wider rivers, *booms and barriers* may be used to keep waterways clear of weeds.

Due to fragmentation of the plants and the risk of infestation of previously uninfested water bodies, **mechanical control** should probably be avoided if possible. Any fragments produced during removal will be carried downstream to potentially infest new water bodies. However, in some situations *mechanical harvesting* is performed.

Biological control agents can be used in rivers and streams if the agents are adapted to living in such conditions. The release of introduced species into these areas probably should be avoided as the spread will be hard to track in flowing water. *Herbivorous insects* that are approved biological control agents could be used, and the flowing water could even provide an advantage by moving individuals to other hydrilla-infested areas. *Triploid Asian grass carp* cannot be used in streams and rivers; although these carp are sterile, it is not permitted to release them into open water bodies unless precautions are taken to restrict the fish to certain areas. Asian grass carp are long lived and will consume other types of vegetation if they end up in a water body without hydrilla. It is recommended that Asian grass carp should only be released in closed water bodies.

In rivers and other flowing water bodies, **chemical control** can be difficult as the water flow quickly diminishes the concentration of herbicide active ingredients in the treated area. Additionally, the water flow increases the possibility that non-target areas and organisms are exposed to potential hazards of applied herbicides. Herbicide residues in runoff water must always be below the threshold levels allowed for the uses of that water. In general, the water flow makes repeated treatments necessary.

Management of Submersed Weeds in Canals

Aquatic vegetation is frequently an issue in canals. The presence of any aquatic vegetation in irrigation systems such as canals affects the water flow and limits its use. One aspect of canals that provides for more management options is the fact that they are manmade so **preventive control** through smart design can be applied *during the construction* of canals. Banks with a steep slope offer little area with shallow water (less than 2 to 3 feet deep) where aquatic weeds would become established. Furthermore, leveling and smoothing of the banks will eliminate areas that otherwise might be hard to reach when weeds need to be removed.

Herbicide — a substance that kills weeds (usually a chemical compound)

Triploid [adj.], triploidy [n.] — when an organism has three sets of chromosomes; this is a rare condition in nature and leads to sterility in most organisms



Figure 41. Surface mats of hydrilla in Wacissa River Springs, Florida. Photograph by Verena Lietze, University of Florida.



Herbicide — a substance that kills weeds (usually a chemical compound)

Herbivorous [adj.], herbivory [n.] — plant-eating

Terrestrial — relating to the earth, for example, an organism that lives on land (as opposed to one that lives in water)

As a **physical control**, lining of the canals (i.e., creating *benthic barriers*) can help prevent initial establishment of weed populations. **Be advised that in Florida, it is illegal to cover large underwater areas, because it is possible that subterranean gas formations accumulate and may lead to dangerous eruptions.** Other physical control methods such as *hand pulling* and *suction harvesting* may be used but should be left to experts. Canals are frequently deep with steep sides and fast flowing water so conditions are not easy for manual plant removal. One person working in an 18 feet wide canal can hand pull vegetation at a rate of approximately 15 feet per hour.

Chaining is used in canals that have access to both sides. As always with mechanical harvesting it is important to collect all fragmented plant material. Aquatic plants grow best in sediment that is 10-20% organic matter. Removal of the top layer of sediment by *dredging* typically removes organic matter making fewer nutrients available for plant growth. Dredging also removes tubers along with the sediment. In some cases the sediment can be removed to below the depth that plants can grow. For hydrilla this is unlikely to be effective as hydrilla can grow in very deep water, it has been found at a depth of 45 feet in Crystal River, Florida!

Drawdowns are applicable in canals with a dam or water control structure. As with other water bodies, drawdowns should be completed in the winter. If canals are not used for navigation, physical control with *boom barriers* also is an effective means to prevent clogging of water control structures.

Mechanical control with *harvesters* or *drag lines* can be used in canals. Harvesters used in canals are usually tractor mounted and operate from the bank with a cutting bar on a hydraulic boom.

Biological control using *Asian grass carp* is an option provided the water body is closed. Screens may be installed to prevent movement into open areas. As in irrigation canals, any vegetation can be a problem for the utility of the water body then Asian grass carp are a great option as they will eventually remove all of the vegetation from the canal and keep it that way for many years. One problem with Asian grass carp is that they may cluster in certain areas of the canals. They prefer earthen canals to concrete lined canals and will leave shallow areas of depth less than one meter so the control provided may not be even throughout the system. Additionally, if the carp manage to remove all the vegetation and are hungry they may start to eat terrestrial vegetation such as overhanging plants and turf on the edge of the water body. *Herbivorous insects* may be useful in canals.

Depending on the water movement, **chemical control** options are similar to those in flowing or in static water. It is important to check whether the water from canals will be used for other purposes, such as irrigation or drinking water supply for humans or other animals. In these cases, temporary or permanent restrictions may not allow the use of certain herbicides.

Management of Submersed Weeds in Static Water

Lakes, ponds and reservoirs are the water bodies that most commonly suffer with vegetation problems. Excessive vegetation prevents recreational use, such as fishing, swimming, and boating. In summer and winter, extremes in temperature cause plant material to die off, which results in an oxygen shortage. This oxygen shortage, if extreme enough, may result in fish kills.

Preventive control can be applied *during the construction* of ditches and ponds. If the banks are designed with a steep slope, areas with shallow water (less than 2 to 3 feet deep) where aquatic weeds become established can be reduced easily and substantially. Furthermore, leveled and smoothed banks are easy to access so that weeds can be removed quickly should they begin to grow.



Furthermore, a *fertilization* regimen might help reduce the establishment of rooted aquatic plants because the nutrients will foster the growth of beneficial plankton, which in turn reduces the amount of sunlight available below the water surface. Care should be taken when applying fertilizers to water bodies as excessive nutrients often results in algal blooms and adding fertilizers might be illegal in your area. The type of algae that dominates will depend on the nitrogen to phosphorus ratio. If the level of nitrogen is low, then blue-green algae thrive; if the level of phosphorus is low, then green algae dominate. Green algae are better for the ecosystem than blue-green algae as they are a food source for many organisms.

Of the **physical control** tactics, *drawdowns in combination with removal of the roots* (including tubers) of hydrilla can be effective in lakes and ponds that allow manipulation of the water level. Drawdowns should be conducted in late fall and last throughout winter. *Manual removal* is a feasible method near the shore line when small amounts of plant material during early infestations are to be removed. Shallow ponds and lakes may allow for effective *dredging*.

The success of **mechanical control** depends greatly on the accessibility of the infested area, which is unique to almost every lake or pond. Even though certain equipment may perform well in one habitat, it may not be suitable in another. To increase the chance of long-term control, *mechanical harvesting* should best be followed by additional methods, such as the introduction of biological control agents.

The *Asian grass carp* has been used very successfully as a **biological control** tactic to reduce and manage hydrilla infestations in lakes and ponds (Figure 42). It is important to prevent the fish from moving to other water bodies, so inlets and outlets must be secured by fences or gates. You can find details and important considerations on the use of grass carp for biological control of hydrilla on pages 39-43.

Chemical control of submersed weeds, such as hydrilla, in static water or water with slow movement can best be conducted by surface spraying, injection into the water column, or application of granules.

Surface and injection treatments apply the herbicide as concentrate with specialized delivery systems. These systems are very effective because they are calibrated appropriately to deliver the correct amount of herbicide and use GPS units to guide the application equipment.

Granular products are most effective when applied evenly across the water surface. The granules will sink to the bottom, where they target the submersed weeds and, through slow-release formulations, provide long-term exposure of the weed to the chemical.

Options for **integrated management** in lakes and ponds are numerous and greatly depend on the specific situation found in the infested habitat. To develop a successful customized management plan, consult with aquatic weed specialists in your area.

Management of Submersed Weeds in Large Impoundments

Reservoirs that are used for recreation and water storage are likely to become infested with hydrilla due to the frequent introduction of boats that may carry attached hydrilla fragments from use at a previous location.

Physical control is possible in large impoundments. In particular, you can take advantage of the water control structures in these artificial water bodies and perform a *drawdown* (Figure 43). Exposing just the shallow areas where most plant material will be growing will enable fish and other aquatic animals to survive during the process. As with all other water bodies, a drawdown should be done in the winter. *Barriers or booms* may be used to

Habitat — an area or environment where a plant, fungus, or animal normally lives and grows

Submersed — a plant with most leaves growing underwater; flowers and some of the leaves may float on the water surface

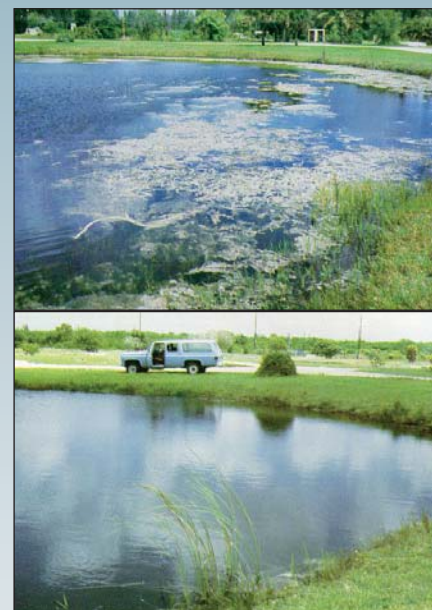


Figure 42. A pond in southeast Florida with a heavy infestation by hydrilla before management (top) and one year after stocking the pond with Asian grass carp at 40 carp per acre (bottom). Photograph by David Sutton (retired), University of Florida.



Herbicide — a substance that kills weeds (usually a chemical compound)

restrict the weeds to certain areas and prevent disruption of activities or use of the water body. *Dredging* may be used, particularly in artificially constructed impoundments, to remove the nutrient-rich sediment and reduce the suitability of the soil for plant growth.

Mechanical control methods can be particularly helpful when the water body is used for drinking water or livestock watering and herbicide use may be undesirable. *Mechanical harvesting* removes the vegetation and so reduces the nutrient load on the system and also does not entail any post-treatment restrictions.

When suitable **biological control** agents are available, these can be used effectively in dammed rivers and other impoundments. *Asian grass carp* have effectively controlled hydrilla in reservoirs. When introducing grass carp, you must ensure that any access to open water bodies is blocked by screen of the correct size to prevent grass carp exit. Incremental stocking, which means starting at low numbers of fish and observing the level of control before adding more, may even permit some submerged aquatic plants to remain.

Chemical weed control that is found effective in lakes and ponds often performs poorly in large impoundments because of the differences in water flow and current. To overcome this difficulty, it can be helpful to apply the maximum recommended rate, select fast-acting herbicides (see Chemical Control section on pages 51-57, 63), use granular formulations, conduct bottom (injection) treatments, or conduct a spray treatment during times with minimum wind. If the impoundment is used for irrigation, make sure to apply only those herbicides that are labeled for such use and observe the post-treatment use restrictions.



Figure 43. Drawdown exposing hydrilla on Rodman Reservoir, Florida. Photograph by Jeff Schardt, Florida Department of Environmental Protection.

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Chapter 6



Insects and Fish Associated with Hydrilla

Contributing Authors
Listed with Each Section

CONTENTS

Hydrilla Leafcutter Moth	78
Hydrilla Leaf-mining Flies	83
Hydrilla Fly Parasitic Wasp	90
Hydrilla Tip Mining Midge	96
Hydrilla Stem Weevil	101
Hydrilla Tuber Weevil	106
Waterlily Leafcutter	111
Asian Grass Carp	117

Herbicide — a substance that kills weeds (usually a chemical compound)



The articles on the next pages were originally published as a Featured Creatures article by the UF/IFAS Entomology and Nematology Department. Check for updates online. URL: <http://entnemdept.ufl.edu/creatures>
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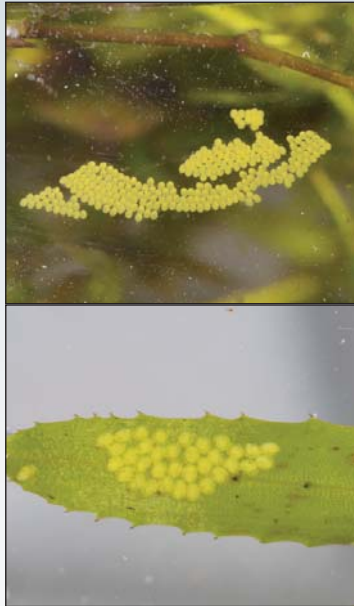


Figure 44. Eggs of *Parapoynx diminutalis* Snellen, within one day of being laid (top). Egg mass sizes vary and are often laid on plant tissue (bottom). Photographs by Lyle Buss, University of Florida.

This chapter introduces the fish and insect species that have been found associated with hydrilla. You can find specific information on their potential use as biological control agents of hydrilla in the Biological Control section on pages 39-46. Text and images were reprinted in this guide with permission from the authors.

Featured Creatures: The Hydrilla Leafcutter Moth

By Julie Baniszewski, Emma N.I. Weeks, and James P. Cuda

SUMMARY: This article focuses on the hydrilla leafcutter moth. Despite the name, the hydrilla leafcutter moth is a generalist herbivore. The moth was considered for importation but was later found to already be present in Florida. The route of introduction is not known. The online version of this article is available on the UF/IFAS Entomology and Nematology department website Featured Creatures. URL: http://entnemdept.ufl.edu/creatures/BENEFICIAL/Parapoynx_diminutalis.htm

COMMON NAME: hydrilla leafcutter moth (unofficial)

SCIENTIFIC NAME: *Parapoynx diminutalis* Snellen (Insecta: Lepidoptera: Crambidae)

Introduction

Parapoynx diminutalis Snellen is an adventive Asian moth with an aquatic larval stage. The moth is found associated with a variety of water bodies including river backwaters, lakes, and ponds (Habeck 1996). The aquatic larvae commonly attack hydrilla, *Hydrilla verticillata* (L.f. Royle), and other aquatic plants (Buckingham and Bennett 1989, 1996).

The moth was identified in 1971 in India and Pakistan during scouting trips to attempt to determine potential biological control agents for hydrilla. Despite having potential for hydrilla destruction, the moth was declared to be a generalist feeder and unsuitable for release into U.S. water bodies for hydrilla control (Baloch et al. 1980). However, the moth was later found in Florida in 1976 by United States Department of Agriculture technicians who were testing herbicides for hydrilla control. The larvae (caterpillars) found on hydrilla were observed to be eating the invasive weed. The pathway, method, or time of the moth's arrival remains unknown (Del Fosse et al. 1976).

Synonymy

According to the global Pyraloidea database (Nuss et al. 2003-2013) and Shibuya (1928) the following junior synonyms have been used for *Parapoynx diminutalis*:

Parapoynx dicentra Meyrick, 1885

Oligostigma pallida Butler, 1886

Nymphula diminutalis Meyrick, 1894

Nymphula uxoralis Strand, 1919

Distribution

Parapoynx diminutalis is native to parts of Asia, Africa, and Australia (Buckingham and Bennett 1996). In its adventive range, *Parapoynx diminutalis* has been found in Panama (notably in the Panama Canal, which was infested with hydrilla), Honduras, and Florida. In commercial greenhouses, the moth has been observed colonizing aquatic plants in England and Denmark (Agassiz 1978, 1981).

Parapoynx diminutalis was first seen in Florida in Fort Lauderdale in 1976 but progressively appeared in more northern counties, eventually reaching Alachua and Putnam counties by 1979 (Balciunas and Habeck 1981). In the early 1980s, hydrilla surveys in other southeastern states revealed that the moth's range did not extend beyond Florida (Balciunas and Minno 1985).

Even in northern Florida, the cooler water temperatures caused populations to be reduced in late winter and early spring. Milder climates such as those found in Panama may enable populations to thrive throughout the year (Buckingham and Bennett 1996). More recent studies indicate that the moth also is established in Louisiana, where 37 moths were collected from 1984-1992 (Brou Jr. 1993).

Description

EGGS: Eggs are smooth and bright yellow when laid (Figure 44); they turn white, and then become transparent as they develop. The eggs are generally deposited on plant leaves or stems just below the water surface in masses of various sizes (Buckingham and Bennett 1996).

LARVAE: Larvae can be distinguished from those of other aquatic species by the presence of branched gills (Habeck 1996) and brown spots on the head and the tip of the thorax. The larval stage consists of seven instars; all seven are off-white with yellow-brown legs (Habeck and Balciunas 2005). The larvae are mobile and feed on hydrilla leaves.

The first instar is white yet nearly transparent and has 1 mm long setae (hairs) (Figure 45). Instars 2 through 7 are white, later instars begin to turn yellow as they approach pupation (Figure 46). In later instars, the length increases and the external gills develop (Figure 47). Instars 3 through 7 use plant tissue to construct a silk case, and often retreat into the case between feeding events (Buckingham and Bennett 1996).

PUPAE: Pupae have three tubercles (or nodules) along each side and two setae (or hairs) on the head. Female pupae can be distinguished from males by their larger size and by their antennae. Female antennae are shorter, extending only to the wing tips, whereas male antennae are longer and extend past the wing tips (Buckingham and Bennett 1996).

Late seventh instar larvae (or pre-pupae) enclose themselves in a white cocoon, which is attached to a submerged plant stem (Figure 48). After 1-2 days in the cocoon, the white pre-pupae have developed into yellow pupae inside the cocoon. The eyes turn red, then brown, and the wings become visible as pupation progresses (Buckingham and Bennett 1996).

ADULTS: Moth adults are white with brown or tan markings or bands on the wings and tan bands on the body (Figure 49). Females typically differ from males by their longer wingspans, more pointed forewings, larger abdomens and shorter antennae, and they lack the noticeable white setae displayed by males on the tip of the abdomen (Buckingham and Bennett 1996).

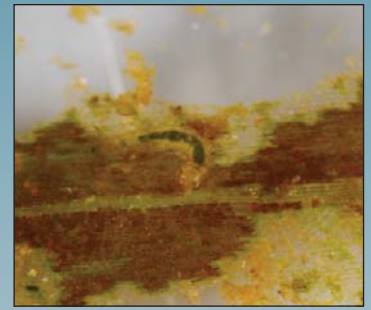


Figure 45. *Parapoynx diminutalis* Snellen, first instar larva eating hydrilla. First instar larvae are transparent, allowing consumed hydrilla to be visible in the gut. Photograph by Lyle Buss, University of Florida.



Figure 46. An early instar of *Parapoynx diminutalis* Snellen (top). Larvae are mobile and retreat into a cocoon between feedings. Cocoons are constructed of plant materials and attached to a hydrilla stem (bottom). Photographs by Julie Baniszewski, University of Florida.

Life Cycle and Biology

Parapoynx diminutalis undergoes complete metamorphosis from an aquatic caterpillar to a moth. Life stages include the egg, seven larval instars (the seventh instar includes a pre-pupa stage), the pupa and finally adult emergence (Buckingham and Bennett 1996). The life cycle of *Parapoynx diminutalis* ranges from 25 to 41 days for development and about five days for the adult life span.

Females lay on average about 200 eggs, but can lay just a few to over 500. The eggs require 4-6 days to develop before first instars hatch. Adults typically emerge from pupae after dusk and are quick to fly to avoid potential predators. The adults drink water using a reduced proboscis, but they do not appear to feed (Buckingham and Bennett 1996).

Parapoynx diminutalis mating has not been studied in detail but has been observed occasionally and seems to occur at around three hours after dusk. Although the maximum time *in copula* is unknown, pairs of moths *in copula* facing in opposite directions were noted to rest for at least 30 minutes. After mating, there is a 1-day pre-oviposition period. Females then oviposit soon after dusk just below the water surface on leaves or stems. First instars have been shown to hatch both below and above the water surface, although it has been observed that the females typically oviposit below the water surface (Buckingham and Bennett 1996).

Hosts

Larvae are commonly found on the aquatic weed hydrilla, *Hydrilla verticillata*. The initial discovery of the moth on hydrilla led to an interest in the moth as a possible biological control agent of this invasive weed. In the field, larvae and pupae have been found in small numbers on coontail (*Ceratophyllum demersum* L.), southern naiad (*Najas guadalupensis* (Sprengel) Magnus), and Illinois pondweed (*Potamogeton illinoensis* Morong) (Buckingham and Bennett 1996).

Furthermore, in laboratory studies, while *Parapoynx diminutalis* larvae preferred hydrilla, they could also complete development on various other plants including coontail, southern naiad, fanwort (*Cabomba caroliniana* Gray), Brazilian waterweed (*Egeria densa* Planchon), and Eurasian watermilfoil (*Myriophyllum spicatum* L.) (Buckingham and Bennett 1989).

Damage

Plant damage is inflicted by the larvae, which not only eat the leaf and stem tissue, but use these materials to prepare their pupal cocoon as well (Figure 50). The main food source for *Parapoynx diminutalis* is the aquatic weed hydrilla (Buckingham and Bennett 1996). In most natural situations in the U.S., hydrilla is invasive and an undesirable weed because it develops surface mats and disrupts natural ecosystems (Haller and Sutton 1975; Hetrick and Langeland 2012).

There have been few studies to quantify the effect of feeding moth larvae on hydrilla biomass. Feeding of the moth larvae on hydrilla in Florida was thought to have a positive effect on hydrilla-invaded water bodies (Del Fosse et al. 1976) by reducing the need for herbicide applications to control hydrilla mats. However, naturalized populations of this moth are too sporadic to have a significant effect on hydrilla density. For example, in northern Florida populations build up during the summer months and can cause extensive defoliation of hydrilla, but in the winter, populations decline rapidly with cooler water temperatures (Buckingham and Bennett 1996).



Figure 47. A late instar of *Parapoynx diminutalis* Snellen, feeding on hydrilla (top). Instars 2 through 7 are white, later instars begin to turn yellow closer to pupation (bottom). Photographs by Lyle Buss, University of Florida.

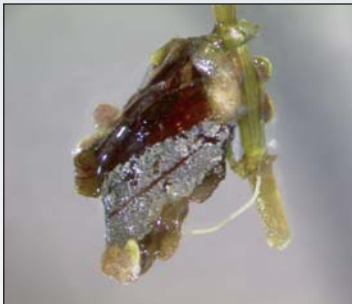


Figure 48. A cocoon constructed and occupied by a *Parapoynx diminutalis* Snellen larva. Photograph by Julie Baniszewski, University of Florida.

Importance as a Biological Control Agent

The moth was identified in Pakistan and India during scouting trips to locate potential biological control agents for hydrilla (Baloch et al. 1980). At this point, the researchers observed the moth damaging hydrilla and believed that the moth could be an effective control agent due to its destructive capabilities.

An important characteristic of a biocontrol agent is host specificity. When laying eggs, female moths are not highly selective, which makes other plants susceptible to consumption by developing larvae. Furthermore, the moth is limited by winter temperatures, and populations decline during the cooler months to a level that is almost undetectable. Sensitivity to cooler climates and lack of host specificity makes the moth a poor biological control agent of hydrilla (Habeck and Balciunas 1976, Buckingham and Bennett 1989).

Monitoring and Management

Monitoring for adult moths can be done using ultraviolet (UV) black lights or incandescent light bulbs, which are both attractive to the moth (Buckingham and Bennett 1996).

Hydrilla is invasive, and the actions of the moth rarely require management and are usually considered to be desirable. However, in certain situations where the presence of hydrilla is needed, such as in research with other biocontrol agents, management of the moth larvae may be necessary to prevent consumption of the plant material. In these situations, a strain of the biorational insecticide *Bacillus thuringiensis* has been tested for controlling *Parapoynx diminutalis*.

Bacillus thuringiensis subspecies *kurstaki*, commonly known as Btk, is specific to lepidopteran pests. Btk produces proteins that are toxic to larvae; the proteins bind to the midgut when consumed and kill the larvae (Bauce et al. 2006, Van Driesche et al. 2008). A commercially available Btk product has been shown to cause 80% mortality of *Parapoynx diminutalis* larvae in about four days (Buckingham and Bennett 1996).

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Figure 49. An adult *Parapoynx diminutalis* Snellen, female moth (top) and male moth (bottom). Females have longer wingspans, more pointed forewings, and larger abdomens. Males have longer antennae and more distinct white setae (hairs) at the tip of the abdomen. Photographs by Lyle Buss, University of Florida.

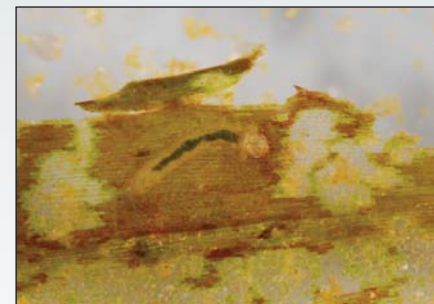


Figure 50. Damage inflicted on hydrilla leaf by a first instar of *Parapoynx diminutalis* Snellen. Photograph by Lyle Buss, University of Florida.



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Featured Creatures: The Hydrilla Leaf-mining Flies

By Emma N.I. Weeks, James P. Cuda, and Jennifer Russell

SUMMARY: This article focuses on the hydrilla leaf-mining flies, *Hydrellia* species. The genera include several species that were imported and introduced for biological control of hydrilla. The online version of this article is available on the UF/IFAS Entomology and Nematology department website Featured Creatures. URL: http://entnemdept.ufl.edu/creatures/BENEFICIAL/hydrilla_leafmining_flies.html

COMMON NAME: hydrilla leaf-mining flies (unofficial)

SCIENTIFIC NAME: *Hydrellia* spp. (Insecta: Diptera: Ephydriidae)

Introduction

Several native and introduced species of flies in the genus *Hydrellia* (Figure 51) are important because they feed on hydrilla (*Hydrilla verticillata* L.f. Royle), an invasive aquatic plant that has been classified as a Federal Noxious Weed. Hydrilla has invaded aquatic ecosystems in Florida and across the U.S. Larvae of *Hydrellia* spp. mine the leaves of hydrilla.

In Florida, there are four species that have been associated with the invasive aquatic weed hydrilla: two native species and two species that were introduced for biological control of hydrilla. The native species are *Hydrellia bilobifera* Cresson and *Hydrellia discursa* Deonier. The introduced species are *Hydrellia pakistanae* Deonier and *Hydrellia balciunasi* Bock.

Distribution

Hydrellia bilobifera is known to feed on hydrilla (Balciunas and Minno 1985), although it also feeds on other aquatic weeds (Center et al. 1998). Although it is not known if *Hydrellia discursa* also feeds on hydrilla, the species is found often in association with hydrilla so the weed is likely to act as a food source (Cofrancesco et al. 2005c). In contrast to the introduced species, *Hydrellia bilobifera* causes minimal leaf damage but mines the stems.

The Asian hydrilla leaf-mining fly, *Hydrellia pakistanae*, was introduced to Florida first in 1987 after being collected in India in 1985. The native range of *Hydrellia pakistanae* includes India, Pakistan and China (McCann et al. 1996). The Australian hydrilla leaf-mining fly, *Hydrellia balciunasi*, was introduced to Florida in 1989 after being collected in Australia in 1982 and taken to Florida for evaluation as a biological control agent and for mass rearing. Releases of *Hydrellia pakistanae* resulted in successful establishment of the fly in Florida as well as in other southeastern states (Cofrancesco et al. 2005a).

Hydrellia balciunasi failed to establish in Florida and is found in low numbers in only a couple of water bodies in Texas (Cofrancesco et al. 2005b). Thanks to natural dispersal of the flies, most of the hydrilla-infested water bodies in Florida have established populations of *Hydrellia pakistanae*. However, hydrilla is still a serious problem in many water bodies. The lack of control provided by *Hydrellia pakistanae* in these instances is likely due to low densities in the field as a result of various abiotic and biotic factors (Wheeler and Center 2001, Cuda et al. 2008) such as parasitism by the native wasp *Trichopria columbiana* (Ashmead).



Figure 51. Adult of hydrilla leaf-mining fly, *Hydrellia* spp. Photograph by Lyle Buss, University of Florida.

Description

EGGS: Eggs are glossy, white, around 0.5 mm in length and look similar to a grain of rice (Figure 52). Under magnification, the eggs exhibit longitudinal ridges, which are characteristic of the genus. Eggs are laid individually on hydrilla or other aquatic vegetation, and a female may lay hundreds of eggs throughout her life span (Cofrancesco et al. 2005a).

LARVAE: Larvae are white, soft bodied and difficult to differentiate among species (Cofrancesco et al. 2005c). There are three instars. Each instar increases in size from around 0.5 to 2 mm in length and 0.2 to 0.5 mm wide. The color changes depending upon nutritional status; feeding larvae are often green due to chlorophyll in the hemolymph. After hatching, larvae burrow into the plant tissue of the hydrilla leaf, forming burrows or mines (Figure 53).

The mouthparts consist of a pair of hooks to rasp away the inner tissue of the leaf (Cofrancesco et al. 2005c). Each larva will feed on about 12 leaves before it is ready to pupate (Cofrancesco et al. 2005a). When it is ready to pupate, the late third instar uses its needle-like modified spiracular peritremes (i.e., the area around the spiracle) to pierce the tissue of the stem. This attachment provides oxygen to the pupae (Deonier 1971).

PUPAE: Pupae are found within a cigar-shaped puparium that is formed from the last larval cuticle and looks like the leaf bud of hydrilla (Figure 54; Grodowitz et al. 1997). The puparia are about 28 mm long and 0.75 mm wide and initially are transparent. At this point, the puparia appear green in color, due to the plant material ingested by the larvae. As the pupa develops, the puparium turns from green to brown. The puparium is attached to a leaf axil and is filled with air.

ADULTS: The adult emerges from the puparium and floats to the water's surface in an air bubble. In *Hydrellia balciunasi*, this occurs between 2 and 4 pm (Buckingham et al. 1991). The adult flies are about 1.5 mm to 2 mm in length and resemble small house flies (Figure 51). The adults have shiny golden heads with green eyes, dark antennae and light-colored maxillary palps. Males can be distinguished from the females by the presence of two lobes at the end of the abdomen (Center et al. 1998).

The two introduced species are not strong fliers and prefer hopping across the water surface to rest on emergent vegetation. The introduced species also are difficult to separate and differentiate from the native *Hydrellia* species. However, there are slight differences in the adult genitalia that a trained professional can distinguish (Grodowitz et al. 1993).

Life Cycle and Biology

Hydrellia spp. undergo complete metamorphosis. Life stages include the egg, three larval instars, the pupa and the adult. After 3 to 4 days, the larvae hatch from the eggs. The larval stage lasts about two weeks before pupation and the development time depends upon the ambient temperature. Pupae develop within the puparium over a period of 6 to 15 days, and adults live approximately 10 to 20 days. Adults of *Hydrellia balciunasi* mate within two hours of emergence (Buckingham et al. 1991). The entire life cycle of *Hydrellia* spp. ranges from 25 to 30 days. *Hydrellia pakistanae* overwinters as first and second instars in hydrilla stems near the water surface (Harms and Grodowitz 2011).



Figure 52. Egg of hydrilla leaf-mining fly, *Hydrellia* spp. Photograph by Lyle Buss, University of Florida.



Figure 53. Larva of hydrilla leaf-mining fly, *Hydrellia* spp. Photograph by Lyle Buss, University of Florida.



Figure 54. Puparium of hydrilla leaf-mining fly, *Hydrellia* spp., puparium containing pupa (top) and empty puparium (bottom). Photograph by Lyle Buss, University of Florida.

Hosts

The imported hydrilla leaf-mining flies were first collected from hydrilla in their respective countries of origin. Laboratory studies and field surveys demonstrated that both imported hydrilla leaf mining fly species are specific to hydrilla. Although the females may lay their eggs on other aquatic plants, larvae prefer to feed on hydrilla.

In host-specificity testing, *Hydrellia pakistanae* females oviposited on all 29 plant species as well as inert objects but preferred hydrilla (Buckingham et al. 1989). Larvae developed on five out of 51 plant species tested but only in low numbers (Buckingham et al. 1989). Curly leaf pondweed (*Potamogeton crispus* L.) supported larval development and produced the most adults but did not sustain subsequent generations (Buckingham et al. 1989). Of 19 aquatic plants examined from its native range in Pakistan, *Hydrellia pakistanae* developed only on hydrilla and *Potamogeton* spp., including *Potamogeton crispus*, *Potamogeton indicus* Roxb. and *Potamogeton perfoliatus* L. (Baloch et al. 1980). Development on *Potamogeton* spp. was limited, and larvae moved onto hydrilla if given the choice (Baloch et al. 1980).

In contrast, *Hydrellia balciunasi* appears to be more host specific. Only one other plant species (*Potamogeton crispus*) out of 14 in a no-choice test and out of 27 in a multi-choice test supported larval development (Buckingham et al. 1991). In the field in its native range in Australia, larvae were found on 24 aquatic plant species, but 97% of the specimens were collected from hydrilla (Balciunas and Burrows 1996).

Damage

Larvae of the hydrilla leaf-mining fly are the damaging stage. Larvae are leaf miners that burrow into the leaf and feed on the inner tissue of the leaf. The mining removes almost all the leaf tissue resulting in transparent leaves (Figure 55). *Hydrellia pakistanae* has been shown to reduce total biomass (30% reduction), tuber biomass (60% reduction) and tuber number (55% reduction) in tank tests (Doyle et al. 2002).

Although each fly larva will consume only about 12 leaves, the reduction in photosynthetic capability (Doyle et al. 2002) and the increased chance of pathogen transmission (Balciunas et al. 2002) are detrimental, especially when the larvae feed in great numbers. A high level of damage by the feeding larvae (70-90%) results in approximately 60% loss in photosynthetic capacity.

The reduction in leaf area also reduces the buoyancy of the plant, and when larval feeding damage is high the plant will die and sink. Patches of hydrilla with intense feeding damage turn brown (Cofrancesco et al. 2005a). In most natural aquatic ecosystems in the U.S., hydrilla is considered to be undesirable either for aesthetic or practical reasons, and tremendous effort and resources are directed toward controlling the plant. Feeding damage by *Hydrellia pakistanae* is thought to have a significant impact on hydrilla biomass and tuber density.

Therefore, many releases have been made across the U.S. in an attempt to control this invasive plant without the need for excessive herbicide applications. However, the density of flies at field sites usually remains low, resulting in limited damage and control, perhaps due to population declines during the winter as well as other factors, such as predation and parasitism (Wheeler and Center 2001, Cuda et al. 2008).



Figure 55. Damage to a hydrilla leaf caused by larvae of hydrilla leaf-mining fly, *Hydrellia* spp. Photograph by Lyle Buss, University of Florida.



Combining the fly releases with other treatments as part of an integrated pest management program is likely to be more effective than the fly damage alone at reducing hydrilla biomass. A combination of herbivory by *Hydrellia pakistanae* and a plant disease-causing pathogen may facilitate plant pathogen transmission and increase plant damage and could provide a sustainable alternative to conventional herbicide-only control efforts. Studies have demonstrated compatibility of the hydrilla leaf mining fly with other control organisms, such as plant pathogens isolated from hydrilla (Shabana et al. 2003).

Importance as a Biological Control Agent

Hydrellia pakistanae is well established in Florida and is found in most water bodies where hydrilla is present. The Asian hydrilla leaf mining fly was collected in India and brought into quarantine in the U.S. in 1985 (Buckingham et al. 1989). Over 3 million individuals were released between 1987 and 1997 in seven states: Florida, Mississippi, Alabama, Louisiana, Georgia, California and Texas (Center et al. 1997). Early releases in Florida between 1987 and 1989 of over 37,000 individuals at 12 sites failed to lead to established populations (Center et al. 1997). Alterations to the rearing and release protocol, including maintaining the colony in the laboratory for a shorter time period and releasing greater numbers of larvae into cages for protection, as opposed to releasing smaller numbers of unprotected eggs, resulted in establishment after later releases (Center et al. 1997).

In southern Florida, 70% of the release sites showed establishment (Center et al. 1997). Dispersal to other sites with hydrilla also has been recorded. For example, high numbers of *Hydrellia pakistanae* were collected in the Miami Canal in 1993, with the nearest release site being over 15 km away (Center et al. 1997). In 1997, *Hydrellia pakistanae* was found to be established from northern Alabama to southern Florida, with records in Texas, Alabama, Georgia and Florida (Center et al. 1997). The effect on hydrilla varies between sites, but as of 1992, 39% of surveyed release sites had well established populations and substantial declines in hydrilla (Grodowitz et al. 1993). However, a decline in hydrilla also was observed in 36% of surveyed sites that had no or limited establishment.

Hydrellia balciunasi, the Australian hydrilla leaf-mining fly, was collected in Queensland, Australia, in 1985 and brought into quarantine in the U.S. in 1988 (Balciunas and Burrows 1996, Grodowitz et al. 1997). It was hoped that, being from Australia, *Hydrellia balciunasi* would be better suited to the climate in the U.S. than *Hydrellia pakistanae* and establish further north (Buckingham et al. 1988). The species was released in Florida in 1989 and in Texas in 1991 (Grodowitz et al. 1997). Between 1989 and 1997, over 280,000 individuals were released at seven sites in Florida and four in Texas (Grodowitz et al. 1997).

In 1997, a survey recorded definite establishment (adults collected for four consecutive months) at only one site in Texas, Sheldon Reservoir (Harris County), and none in Florida (Grodowitz et al. 1997). The site corresponded to the site with the second highest releases (76,000 individuals), whereas the most released at any one site in Florida was 20,000 individuals (Grodowitz et al. 1997). Despite many releases at that site, the numbers of adults remained low and only 5% damage to hydrilla was recorded at peak fly densities in 1992 (Grodowitz et al. 1997). Failure of this insect to establish has been attributed to several factors including competition with *Hydrellia pakistanae*, incompatibility with the U.S. strains of hydrilla, parasitism by the native wasp *Trichopria columbiana* (Ashmead) and inbreeding due to mass rearing (Grodowitz et al. 1997).

Monitoring and Management

Several methods can be used to monitor *Hydrellia* fly activity. Firstly, hydrilla can be collected and the leaves examined for the characteristic damage caused by the larvae (Grodowitz et al. 1997). The larvae and pupae also may be detected under a light microscope. However, this method does not allow for identification to species. To identify the flies to species, it is necessary to either rear the immature stages to adults or collect adults in the field (Grodowitz et al. 1997). One of the best methods to collect adults in the field involves the use of a specially designed hand-held vacuum (Cofrancesci et al. 2005). Berlese funnels also can be used to extract larvae and adults from plant material (Grodowitz et al. 1997, Cofrancesco et al. 2005).

In a few specific situations, presence of hydrilla may be desired, e.g., in ponds or fishing lakes; in these situations, the flies may need to be controlled to prevent damage to the plant. Researchers conducting experiments with hydrilla also may need to chemically or physically exclude *Hydrellia* flies or other hydrilla-attacking insects in order to produce suitable plant material for their investigations.

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Figure 56. Egg of *Trichopria columbiana* (Ashmead). Photograph by Byron Coon, Argosy University.

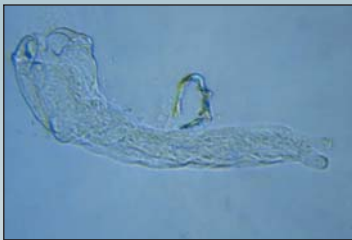


Figure 57. First instar larva of *Trichopria columbiana* (Ashmead). Photograph by Byron Coon, Argosy University.



Figure 58. Second or third instar larva of *Trichopria columbiana* (Ashmead). Photograph by Byron Coon, Argosy University.

Featured Creatures: The Hydrellia Fly Parasitic Wasp

By Byron R. Coon, Nathan E. Harms, Michael J. Grodowitz, Emma N.I. Weeks, and James P. Cuda

SUMMARY: This article focuses on a parasitic wasp of the hydrilla leaf-mining flies, *Hydrellia* species. The hydrilla leaf-mining flies include several species that were imported and introduced for biological control of hydrilla. The online version of this article is available on the UF/IFAS Entomology and Nematology department website Featured Creatures. URL: http://entnemdept.ufl.edu/creatures/misc/wasps/Trichopria_columbiana.htm

COMMON NAME: hydrellia fly parasitic wasp (unofficial)

SCIENTIFIC NAME: *Trichopria columbiana* Ashmead (Insecta: Hymenoptera: Diapriidae)

Introduction

Trichopria columbiana (Ashmead) (Insecta: Hymenoptera: Diapriidae) is a native endoparasitic wasp. The wasp is a parasitoid of *Hydrellia* species (Insecta: Diptera: Ephidridae), with multiple implications to biological control. The hydrellia flies are a diverse group with varied ecological roles. Deonier (1971) described 57 *Hydrellia* species in the Nearctic region. The adults of this genus are semi-aquatic and the immatures are aquatic, feeding on aquatic and semi-aquatic plants (Deonier 1971). Of these 57 species, at least 7 have been described as hosts for *Trichopria columbiana*.

In addition, the wasp has successfully moved from its native hosts to exotic *Hydrellia* species (*Hydrellia pakistanae* Deonier and *Hydrellia balciunasi* Bock) that were imported into Florida for biological control of hydrilla, *Hydrilla verticillata* (L.f.) Royle, which is widely regarded as one of the worst invasive weeds worldwide (Holm et al. 1997). Deonier (1971) reported that *Trichopria columbiana* and other parasitic Hymenoptera can negatively impact population densities of *Hydrellia* spp., especially in certain marginal habitats and when parasitoid population densities are high.

Synonymy

According to Hymenoptera Online, there is only one junior synonym for *Trichopria columbiana* (Johnson 2014):

Diapria columbiana Ashmead 1893

Distribution

Trichopria columbiana is widely distributed in North America (Bennett 2008) and has been reported from the District of Columbia, Virginia (Ashmead 1893), Michigan (Berg 1950), California (Grigarick 1959), Minnesota (Deonier 1971), Alabama (Grodowitz et al. 1997), Florida (Wheeler and Center 2001), and Texas (Doyle et al. 2002).

Description

The description of the life stages of this species has been modified from Coon et al. (in press).

EGGS: Eggs were dissected from *Trichopria columbiana* ovaries to determine their pre-oviposition morphology. *Trichopria columbiana* eggs, which are hymenopteriform (wasp-like) in shape, were 0.19 mm long and 0.06 mm wide. The chorion (outer membrane) is smooth and thin. As the chorion is transparent, the developing embryo is clearly visible (Figure 56). The second inner membrane, which is likely to be the vitelline membrane, is flexible.

A double-membrane egg is characteristic of hydroptic eggs (DeBach 1964). Hydroptic eggs take up nutrients and water from the host's hemolymph for continued development and typically expand in size (Flanders 1950). *Hydrellia pakistanae* pupae were dissected and *Trichopria columbiana* eggs were removed from the host 72 hours post-oviposition. These eggs were much larger than those dissected from the female parasitoid; they measured 0.57 mm in length by 0.28 mm in width.

LARVAE: There are three instars; the first instar is 0.49 mm long and 0.14 mm wide. At this stage, the body is segmented and the mandibles are large and sclerotized (hardened). The end of the abdomen has a two-lobed appendage with several teeth on each lobe (Figure 57). This instar moves freely in the hemolymph of the host and is believed to obtain oxygen by diffusion.

The second instar is 0.92 mm long and 0.31 mm wide, and the third instar is 1.50 mm long and 0.52 mm wide. Both the second and third instars are similar in appearance and are grub-like (Figure 58). The abdominal appendage and large mandibles present on the first instar are absent. The head of the later instars has indistinct mouthparts that are not differentiated from the body. The second and third instars obtain oxygen from the host by attaching to the host tracheal system.

PUPAE: The pupae are enclosed in a thin case (Figure 59), which is believed to be the last larval exuvium (cast skin). The case is transparent and the developing adult is visible inside with the red eyes particularly noticeable. Also visible are many small globules, which are believed to be the fecal material released by the last instar before pupation.

ADULTS: The following description of the adults is based on Ashmead (1893). The overall length of the adult wasp is 1-2 mm. The body is shiny and black in color with the base of the antennae and the legs reddish yellow (Figure 60). The head is round and narrows behind the eyes. The thorax narrows anteriorly forming a round neck. The abdomen is oval shaped. The wings are strongly fringed and are clear with pale yellow veins.

Male and female *Trichopria columbiana* can be distinguished easily by differences in the shape of the antennae. The antennae of females have 12 segments and are slightly clavate or club-like. In contrast, the antennae of males have 13 segments and are filiform or thread-like.

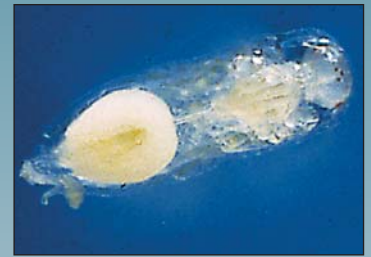


Figure 59. Pupa of *Trichopria columbiana* (Ashmead). Photograph by Byron Coon, Argosy University.



Figure 60. Adults of *Trichopria columbiana* Ashmead; male (A) with filiform or thread-like antennae and female (B) with slightly clavate or club-like antennae. Photographs by Nathan Harms, U.S. Army Engineer Research and Development Center.

Life Cycle and Biology

The life cycle and biology was studied in detail by Coon et al. (in press) and is summarized below. Four behaviors associated with host location were observed in the laboratory:

1) searching, 2) stem examination, 3) oviposition, and 4) grooming and/or resting.

When searching for a host, *Trichopria columbiana* need to be able to access the pupae of *Hydrellia* species, which are usually underwater. The female inserts her antennae into the water first, presumably to detect chemical cues of plant damage or the host insect. When given a choice of hydrilla with *Hydrellia* pupae and hydrilla with *Hydrellia* larvae, 96% of parasitoids selected the hydrilla with *Hydrellia* pupae.

Therefore, a suitable host is located using chemoreception. The adult female wasp swims underwater by trapping a bubble of air under her wings, which she uses to breathe. After locating a suitable host, the female inserts her ovipositor into the thorax of the fly, which is close to the cuticle of the puparium. The female parasitoid prefers to lay her eggs in early- to intermediate-stage pupae. However, eggs may also be laid in late instars (Grodowitz et al. 2009).

Dissections of both *Hydrellia pakistanae* and *Hydrellia balciunasi* revealed that *Trichopria columbiana* deposits a maximum of three eggs per host. Each female may lay 14 to 32 eggs in a lifetime. The female deposits her eggs directly into the host hemolymph, and the eggs develop in 1 to 3 days. Development of the first instar requires 1 to 3 days, and only a single larva survives. Apparently, the surviving larva uses its mandibles to kill siblings, thereby avoiding competition for resources. The estimated stadia lengths (time between molts) for the second and third instars is 2 to 5 and 5 to 8 days, respectively. Development of the larval stage is completed in 13 to 23 days.

The pupal stage of *Trichopria columbiana* lasts between 5 and 7 days. After pupation, the adult parasitoid exits the host, which is usually below the water surface, by cutting a hole in the end of the puparium that is not attached to the tracheal system with its mandibles. The adult parasitoid floats to the surface with an air bubble attached to hairs on its abdomen. The bubble of air is believed to have been acquired from the internal environment of the host puparium. This assumption is based on the fact that *Hydrellia* fly adults exit their puparia in a similar way but ascend to the surface enclosed within an air bubble obtained from inside their puparium (Balciunas et al. 2002).

Total development time from egg to adult was on average 22 days (14 to 26 days) in the laboratory at 25 °C. *Trichopria columbiana* overwinters as an adult in hydrilla at the edge of the water body. The sex ratios that have been recorded in Florida and Texas are female-biased with males being relatively rare. Collection of adults from hydrilla resulted in 14,776 individuals, of which only four were male—a sex ratio of 1: 3,694 (male: female).

Hosts

According to Deonier (1971), *Trichopria columbiana* attacks at least seven native *Hydrellia* spp. including *Hydrellia ascita* Cresson, *Hydrellia bergi* Cresson, *Hydrellia cruralis* Coquillett, *Hydrellia griseola* (Fallén), *Hydrellia ischiaca* Loew, *Hydrellia luctuosa* Cresson, and *Hydrellia pulla* Cresson. This parasitoid also attacks the two *Hydrellia* spp., *Hydrellia pakistanae* (Cuda et al. 1997) and *Hydrellia balciunasi* (Grodowitz et al. 1997), that were introduced in the U.S. for biological control of the aquatic weed hydrilla.

Damage

The parasitoid lays eggs in the pupae of *Hydrellia* spp. Once the larvae hatch and begin feeding, the developing *Hydrellia* pupa provides the food source for the larvae. The developing pupa is killed and will not develop into an adult fly. Therefore, *Trichopria columbiana* can reduce populations of *Hydrellia* species including hydrilla leaf-mining flies.

Importance for Biological Control

Trichopria columbiana is a parasitoid of *Hydrellia* fly species. Depending on the ecological role of the host species, *Trichopria columbiana* can have a positive or negative effect on biological control.

Some *Hydrellia* species, including the introduced biological control agents *Hydrellia pakistanae* and *Hydrellia balciunasi*, feed on the invasive aquatic weed hydrilla, *Hydrilla verticillata*. After its introduction into the U.S. by the aquarium industry in the 1950s (Langeland 1996), various control methods, including biological control, were developed and used to manage infestations. Classical biological control studies were initiated in the 1970s (Buckingham 1994). These efforts led to the release of four insects in the U.S., two of which were the leaf-mining ephydrid flies, *Hydrellia pakistanae* and *Hydrellia balciunasi* (Center et al. 1990).

Despite successful establishment and range expansion of the Asian hydrilla leaf-mining fly, *Hydrellia pakistanae*, population levels of the insect and associated plant damage have remained low (Cuda et al. 2008). However, there is some evidence that recent declines of hydrilla in Florida and Texas have been associated with local increases in *Hydrellia* fly populations (Grodowitz et al. 2004). Several abiotic and biotic factors have been identified that could adversely affect *Hydrellia pakistanae* populations on a landscape scale (Cuda et al. 2008). One of the potentially limiting biotic factors is parasitism by the native endoparasitic wasp *Trichopria columbiana*.

In Florida, the highest parasitoid activity was recorded in the cooler winter months, October to January, with a peak in January. The average parasitism rate in Florida and Texas was around 20-30% (Coon et al. in press; Grodowitz et al. 2009). Buckingham and Okrah (1993) concluded that parasitism of the introduced *Hydrellia pakistanae* and *Hydrellia balciunasi* by parasitoids of native *Hydrellia* spp. could be more of a problem than interspecific competition between the two introduced biological control agents. Hence, they suggested that parasitism should be carefully monitored.

In hindsight, attack of the two introduced *Hydrellia* spp. by *Trichopria columbiana* or other parasitoids of native *Hydrellia* spp. was predictable because the biocontrol agents were not released in an 'enemy-free space' (Lawton 1985). When weed biological control practitioners select agents, they should carefully consider the potential for insects to acquire novel parasitoids. This precaution will help avoid reducing biological control agent effectiveness and apparent competition, particularly where species interact through shared natural enemies.

The parasitoid also has been found in *Hydrellia pulla* pupae, with 63% of 61 puparia being parasitized during one summer in Minnesota (Deonier 1971). *Hydrellia pulla* feeds on pondweeds (Deonier 1971), such as the large-leaved pondweed (*Potamogeton amplifolius* Tuck.), variable-leaf pondweed (*Potamogeton gramineus* L.), and Richardson's pondweed (*Potamogeton richardsonii* [Benn.] Rydb.), which are all classified as endangered or threatened in their native ranges in the U.S. For this reason, the parasitoid may protect native pondweeds from herbivory by *Hydrellia*.

On the other hand, *Trichopria columbiana* is a biological control agent itself, providing control of agricultural pests of rice crops. Grigarick (1959) observed 60% parasitism in one sample of *Hydrellia griseola* mining rice plants in California. In that same study, low parasitism of the first generation of *Hydrellia griseola* was observed, but parasitism approached almost 90% in succeeding generations. Deonier (1971) found 38% parasitism by *Trichopria columbiana* and other parasitoids in 132 puparia of *Hydrellia ischiaca*, a pest of wild rice crops.





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(Scan the QR code to connect to the article.)

Monitoring and Management

Adult parasitoids can be extracted from plant material by using Berlese funnels. *Hydrellia* pupae can be dissected or isolated and placed in rearing containers to determine parasitism rates.

Acknowledgements

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Figure 61. Dorsal views of adult female hydrilla tip mining midge, *Cricotopus lebetis* Sublette. Photographs by Jerry Butler, University of Florida.



Figure 62. Dorsal views of adult male hydrilla tip mining midge, *Cricotopus lebetis* Sublette. Photographs by Jerry Butler, University of Florida.

Featured Creatures: The Hydrilla Tip Mining Midge

By James P. Cuda, Byron R. Coon, Emma N.I. Weeks, Judy L. Gillmore and Ted D. Center

SUMMARY: This article focuses on the hydrilla tip mining midge, a hydrilla-eating insect that was discovered in Florida. The route of introduction is not known. The online version of this article is available on the UF/IFAS Entomology and Nematology department website Featured Creatures. URL: http://entnemdept.ufl.edu/creatures/aquatic/hydrilla_tip_mining_midge.htm

COMMON NAME: hydrilla tip mining midge (unofficial)

SCIENTIFIC NAME: *Cricotopus lebetis* Sublette (Insecta: Diptera: Chironomidae)

Introduction

Insects of the family Chironomidae, commonly known as midges, are often the most abundant group of insects inhabiting freshwater environments (Pinder 1986). Midges are fragile and mosquito-like in appearance but they do not bite. Larvae of most midges are aquatic and feed primarily on algae and decaying organic matter. A few species, however, are capable of mining the soft tissues of submersed plants and using the living plant material as a food source (Pinder 1986). Recently, this feeding strategy has been studied in some detail in the genus *Cricotopus* because of the realization that it could be exploited for the biological control of the alien aquatic weed Eurasian watermilfoil, *Myriophyllum spicatum* L. (MacRae et al. 1990) and possibly hydrilla, *Hydrilla verticillata* (L.f.) Royle (Cuda et al. 2002, Cuda et al. 2011).

Hydrilla is a submersed aquatic plant endemic to the Old World tropics; it was introduced into Florida by the aquarium industry in the late 1950s from Sri Lanka (Langeland 1990). After its discovery in the Crystal River watershed in 1960, hydrilla continued to expand its range statewide and to increase in severity in water bodies already infested. The dense surface mats associated with severe hydrilla infestations cause problems because they hinder navigation and flood control, interfere with recreational activities, and reduce the biodiversity in aquatic ecosystems (Haller 1978). Between 1982 and 2013, approximately \$260 million in state and federal funds were spent managing hydrilla in Florida public waters (FWC 2013). In the year 2012-2013, \$7.43 million was spent treating 14,150 acres of hydrilla (FWC 2013). With the recent discovery of herbicide resistance in hydrilla (Michel et al. 2004), there is renewed interest in biological control.

In 1992, USDA researchers discovered midge larvae attacking the apical meristems of hydrilla in the Crystal River watershed in Citrus County, Florida (G.R. Buckingham, personal communication), and that the damaged hydrilla at one site was stunted and unable to grow to the surface. The hydrilla-attacking midge was subsequently identified as *Cricotopus lebetis* Sublette (Figures 61 and 62), a species possibly new to Florida (Epler et al. 2000). Because previous research implicated midge larvae as causal agents of damaged stem tips on stunted hydrilla plants in Africa (Markham 1986), this tip mining midge may have some potential as a biological control agent.

Synonymy

According to NCBI Taxonomy, Species 2000 and the Integrated Taxonomic Information System the following synonyms have been used to describe *Cricotopus lebetis*:

Cricotopus tricinctus Meigen, 1818

Cricotopus hyalinus Kieffer, 1921

Distribution

The midge genus *Cricotopus* is represented in North America by four subgenera (Epler 1995). Two of these subgenera, *Cricotopus* and *Isocladius* occur in Florida and contain at least eight species (Epler 1995). The actual distribution of *Cricotopus lebetis* will not be known with any certainty until it can be determined if it is an immigrant that was accidentally introduced along with hydrilla, or an indigenous species that has developed a new association with hydrilla. However, *Cricotopus lebetis* was found to be widely distributed in Florida from the northern peninsula (Lake Rowell, Bradford Co.) to the south-central part of the state (Lake Istokpoga, Highlands Co.) (Stratman et al. 2013b).

Description

ADULTS: The adult midges are small, only 3 to 4 mm in length, and fragile (Figures 61 and 62). Both sexes are pale green in color with black markings on the thorax and a pair of adjacent dark bands on abdominal segments 2 and 3, and 5 and 6. The black markings on the thorax and the coarse banding pattern on the abdomen give the midge a darker appearance. The sexes can be readily distinguished by the condition of the antennae and the shape of the abdomen. In females, the antennae are short and the abdomen is as wide as the thorax. In contrast, the males possess long antennae with distinct whorls of hair and have a narrow, tapering abdomen.

EGGS: The eggs are laid in a linear-shaped mass, containing from 50 to 250 eggs diagonally-arranged in one or two rows encased in a sticky gelatinous tube (Figure 63). The eggs are white in color when first laid, and resemble a string of pearls. Within 24 hours the eggs that have been fertilized turn grayish-brown, and red eyespots of the fully formed embryo appear just prior to hatching.

LARVAE: The larvae of *Cricotopus lebetis* can be identified by the color and general appearance of the body (Figure 64). Live or freshly-preserved specimens have a characteristic green body color with a broad dark blue band around the thorax. After the body color fades in preserved specimens, the larvae can be separated from other midge larvae by the presence of a pair of lateral setae on each abdominal segment.

PUPAE: The pupae do not feed. The wings and other adult features that have been developing internally are visible (Figure 65). Breathing horns or trumpets that are usually present on the prothorax in species that have free-swimming pupae are lacking. A pupa destined to become an adult female of *Cricotopus lebetis* will have a full complement of eggs visible in the abdomen.



Figure 63. Two views of egg masses of hydrilla tip mining midge, *Cricotopus lebetis* Sublette. Photographs by Jerry Butler, University of Florida.



Figure 64. Larva of hydrilla tip mining midge, *Cricotopus lebetis* Sublette. Photograph by Jerry Butler, University of Florida.



Figure 65. Female pupa of hydrilla tip mining midge, *Cricotopus lebetis* Sublette. Photograph by Jerry Butler, University of Florida.

Life Cycle and Biology

Adult midges live from one to three days and do not feed. They mate on a suitable substrate in daylight (Figure 66). Male swarming behavior that is a prerequisite for mating in many species of the Chironomidae was not observed in this species. Shortly after mating the female lays her eggs on the surface of the water. The female inserts the tip of her abdomen beneath the water surface where she deposits the egg mass and dies soon afterwards. The egg stage lasts 36 to 48 hours.

Larval hatching is synchronous. The neonates are very active but remain inside the tubular gelatinous matrix for several hours, crawling from one end to the other. Eventually, they exit the gelatinous matrix from one end, or occasionally from the middle. The larvae at this stage of their development are free-swimming and vulnerable to predation. However, their translucent color and small size may afford them some protection until they can enter a shoot tip. Larvae can only survive for 48 hours without access to hydrilla so must find a host plant quickly. The larvae complete their development in nine to 22 days.

Pupation occurs inside the hydrilla stem. The pupal stage lasts 24 to 48 hours. Adult emergence occurs after the sedentary pupa exits the stem by undulating its abdomen and slowly swims to the surface aided by an air bubble released inside the pupal skin.

Hosts

The hydrilla tip mining midge was discovered feeding on hydrilla in Crystal River in 1992. One requirement of biological control agents is that they are specific to the target weed. Field collections have so far indicated that *Cricotopus lebetis* is feeding specifically on hydrilla. However, further studies are needed to verify these findings. Laboratory testing revealed that *Cricotopus lebetis* was able to utilize several other species (Stratman et al. 2013a). With no difference in survival compared to hydrilla, *Cricotopus lebetis* developed to the adult stage on three additional aquatic plants: Canadian waterweed, *Elodea canadensis* Michx.; Brazilian waterweed, *Egeria densa* Planch.; and southern naiad, *Najas guadalupensis* (Spreng.) Magnus (Stratman et al. 2013a). Interestingly, monoecious hydrilla was a better developmental host than dioecious hydrilla (Stratman et al. 2013a).

In laboratory choice tests, larvae preferred southern naiad and Canadian waterweed to hydrilla (Stratman et al. 2013a). In laboratory oviposition tests, females preferentially laid eggs on Canadian waterweed, compared to hydrilla, but there was no difference in oviposition preference between southern naiad and hydrilla (Stratman et al. 2013a). The different results between the laboratory and field host specificity observed for *Cricotopus lebetis* can be explained by the fact that for most biological control agents, the laboratory host range is often broader than the field host range.

Damage

The hydrilla tip mining midge feeds on the growing tips of hydrilla plants during the larval stage. Once inside the plant, the larvae mine and feed on the vascular tissues of the apical meristem of the hydrilla shoots (one larva per shoot tip). As they develop to maturity, their feeding activity creates a 1 to 2 cm tunnel inside the stems, which eventually kills the shoot tips and induces their abscission (Figure 67). The tunnels created by the developing larvae inside the shoot tips probably protect them from predators but also function as pupal cases.



Figure 66. Mating adults, hydrilla tip mining midge, *Cricotopus lebetis* Sublette. Male midge with feathery antennae and narrow abdomen on right, female on left. Photograph by Jerry Butler, University of Florida.



Figure 67. Hydrilla tip damage and larva of hydrilla tip mining midge, *Cricotopus lebetis* Sublette. Photograph by Karen Stratman, University of Florida.



Figure 68. Damaged hydrilla tip with pupa of hydrilla tip mining midge, *Cricotopus lebetis* Sublette. Photograph by Jerry Butler, University of Florida.

Before pupating, the mature larva completely severs the tip of the shoot to create an escape route for the fully-developed pupa, and caps the opening of the tunnel with plant fibers excavated from the stem wall (Figure 68). Adult emergence occurs after the sedentary pupa exits the stem by repeatedly undulating its abdomen to break through the fibrous cap. The preparation of the pupal case by the last instar larva is what actually induces abscission of the shoot tip.

Importance as a Biological Control Agent

Cricotopus lebetis may have some potential as a biological control agent of hydrilla. The larvae of this herbivorous midge mine the meristematic tissues of the plant and in the process disrupt shoot growth. By severely damaging or killing the apical meristems, the developing larvae may prevent new stems from reaching the surface thereby changing the plant's architecture. This type of damage is desirable for managing hydrilla because it would eliminate most of the adverse effects caused by the formation of the dense surface mats, such as changes in biodiversity, water chemistry, circulation and temperature.

Efficacy studies have so far produced positive results. A field study conducted in Crystal River, FL, that assessed the hydrilla biomass and compared it to the number of tips damaged by *Cricotopus lebetis* found that with increasing larval midge density there was increasing numbers of tips damaged and a decrease in hydrilla biomass (Cuda et al. 2011). Furthermore, a manipulative experiment conducted in plexiglas tanks in a greenhouse also showed that *Cricotopus lebetis* was able to reduce the biomass of hydrilla by 99% in 2 months (Cuda et al. 2011).

However, except for Lake Rowell in Bradford Co., field populations of *Cricotopus lebetis* are relatively low. For example, a recent study of six lakes in Florida found the midge in only half of the lakes and at low abundance (Stratman et al. 2013b). Augmentation of the population by mass releases could increase the damage to hydrilla and reduce the vegetative biomass at the surface.

Monitoring and Management

Cricotopus lebetis can be monitored in the field by collection of hydrilla and examining the tips for the presence of larvae or larval damage (Cuda et al. 2011). A more reliable method is collecting/holding hydrilla for several weeks in aerated trays within cages and collecting adults by aspirator.

Management is not necessary for the hydrilla tip mining midge as it is not a pest in the U.S. However, it is susceptible to pesticides (Stratman et al. 2013c).

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Featured Creatures: The Hydrilla Stem Weevil

By Emma N.I. Weeks, Michael J. Grodowitz, and James P. Cuda

SUMMARY: This article focuses on the hydrilla stem weevil. The weevil was imported for the biological control of hydrilla. The online version of this article is available on the UF/IFAS Entomology and Nematology department website Featured Creatures. URL: http://entnemdept.ufl.edu/creatures/BENEFICIAL/Bagous_hydrillae.htm

COMMON NAME: hydrilla stem weevil (unofficial)

SCIENTIFIC NAME: *Bagous hydrillae* O'Brien (Insecta: Coleoptera: Curculionidae)

Introduction

Bagous hydrillae O'Brien is a semi-aquatic weevil that feeds on the aquatic invasive weed *Hydrilla verticillata* (L.f.) Royle (Figure 69). Larvae of the weevil mine hydrilla stems and the adults feed on the stems and submerged leaves. This weevil was discovered during overseas surveys for biological control agents for hydrilla during the 1980s and was first introduced to the U.S. in Florida in 1991 after extensive host-specificity testing. Another species, *Bagous affinis* Hustache (Insecta: Coleoptera: Curculionidae), was introduced to the U.S. after being discovered in India, but failed to establish (Cuda and Frank 2013).

Distribution

Bagous hydrillae is native to Australia and was originally collected from 21 sites throughout the Northern Territory and New South Wales. In Australia, higher numbers of weevils were collected from impounded water bodies, compared to rivers and creeks (Balciunas and Purcell 1991).

Description

EGGS: Eggs of *Bagous* species are usually white and the outer membrane or chorion is transparent (Buckingham and Bennett 1994). Eggs of *Bagous hydrillae* are oval and 0.52 mm long by 0.27 mm wide (Figure 70); they are laid by the female inside the hydrilla stem. The female makes a hole in the stem with her mouthparts and then inserts the egg into the hole.

LARVAE: Larvae of *Bagous hydrillae* are relatively non-descript and have not been described in detail in the literature. However, they have the general appearance of weevil larvae in that they are “comma-shaped” grubs with a soft body (Marvaldi 2003). The body appears to be translucent and the gut is visible (Figure 71). There are three instars.

The head capsules of *Bagous* species larvae are light brown and ocelli are present (Gosik 2009, Buckingham and Bennett 1994). The antennae are one-segmented and conical (Gosik 2009). Like other weevil larvae, *Bagous hydrillae* larvae do not have legs. Larvae of the Curculionidae are usually found within dense materials, such as plant tissue (Marvaldi 2003). After hatching, *Bagous hydrillae* larvae bore into the surrounding tissue and are found within tunnels in hydrilla stems.



Figure 69. Adult of the hydrilla stem weevil, *Bagous hydrillae*. Photograph by Gary Buckingham, USDA-ARS.



Figure 70. Exposed egg of the hydrilla stem weevil, *Bagous hydrillae*, inserted into a hydrilla stem. Photograph by U.S. Army Engineer Research and Development Center.



Figure 71. Larvae of the hydrilla stem weevil, *Bagous hydrillae*. Photograph by Gary Buckingham, USDA-ARS.



Figure 72. Pupa of the hydrilla stem weevil, *Bagous hydrillae*. Photograph by U.S. Army Engineer Research and Development Center.



Figure 73. Adult of the hydrilla stem weevil, *Bagous hydrillae*. Photograph by U.S. Army Engineer Research and Development Center.



Figure 74. Adult of the hydrilla stem weevil, *Bagous hydrillae*, moving underwater on plant material. Photograph by Gary Buckingham, USDA-ARS.

PUPAE: When third instars emerge from the stem to pupate, they are white in color but change to yellow. Pupae of *Bagous hydrillae* are referred to as naked as they do not form a protective covering, such as a cocoon (Figure 72). As the pupae approach the end of their developmental period, the eyes darken and the body of the adult becomes apparent.

ADULTS: Adult hydrilla stem weevils are dark brown with lighter spots on the abdomen (Figure 73). Some individuals also have several larger light-colored areas on the elytra (wing covers) and thorax. Males can be distinguished from females by a broad shallow depression on the underside of the abdomen between the last pair of legs; this depression is absent in females (Balciunas and Purcell 1991).

Life Cycle and Biology

Life stages include the egg, larva, pupa and adult. The life cycle from egg to adult takes 12-14 days at 25°C (Balciunas and Purcell 1991). However, the larval development rate can be influenced by hydrilla quality, with decreased time to pupation when hydrilla nitrogen content is high and stems are soft (Wheeler and Center 1997). After a preoviposition period of 6.8 days, females start to lay eggs (Balciunas and Purcell 1991). The female makes a hole in the hydrilla stem with her mouthparts and lays individual eggs near leaf nodes (Balciunas and Purcell 1991). Females lay, on average, three eggs per day and around 100 eggs in a lifetime (Balciunas and Purcell 1991).

Larvae emerge within 54-66 hours (Balciunas and Purcell 1991). Larvae mine the stems of the hydrilla. Around 8 days after oviposition, the third instar emerges from the stem to pupate. Pupation occurs in terrestrial habitats, usually in stranded plant material or in the silt at the edge of the water body. The combined prepupal and pupal period is approximately 6 days (Balciunas and Purcell 1991). Adult weevils are active at night and live for approximately five weeks (Balciunas and Purcell 1991). They move underwater through plant material (Figure 74).

The larvae of a closely related species, the hydrilla tuber weevil, *Bagous affinis*, feeds on hydrilla tubers. Although the female hydrilla stem weevil will lay eggs on tubers and the larvae will develop normally, the opposite is not true. The hydrilla tuber weevil larvae are not able to complete development on stem tissue and die before the prepupal stage (Wheeler and Center 2007a).

Hosts

In laboratory host-range tests, *Bagous hydrillae* appeared to be less host specific than is normally desirable in a biological control agent, feeding to some extent on 16 different plant species (Balciunas et al. 1996). Oviposition and larval survival were evaluated on these 16 plant species, and several other species of interest. *Bagous hydrillae* oviposited on 12/19 species (63%) including hydrilla (Balciunas et al. 1996). Larvae successfully developed to adults in all plant species that were attractive to ovipositing females (Balciunas et al. 1996).

However, in the native range in Australia, hydrilla is the main host for this insect. In total, 1,630 collections of 49 plant species including hydrilla were completed and 90% of the *Bagous hydrillae* adults and 74% of the larvae collected as a result were found to be using hydrilla as a host (Balciunas and Purcell 1991).

In field surveys, weevils were collected from eight of 48 other plant species that were sampled, including an eelgrass species (*Vallisneria gracilis* F.M. Bailey), Brazilian waterweed (*Egeria densa* Planch), duck lettuce (*Ottelia alismoides* [L.] Pers.), coontail (*Certophyllum demersum* L.), a naiad species (*Najas tenuifolia* R. Brown), water snowflake

(*Nymphoides indica*), curly-leaf pondweed (*Potamogeton crispus* L.) and clasping-leaf pondweed (*Potamogeton perfoliatus* L.) (Balciunas and Purcell 1991). However, of the total collections only less than 10% of adults and 26% of larvae were on other plant species. The majority of the weevils not collected from hydrilla (80-85%) were collected from *Vallisneria gracilis* (Balciunas and Purcell 1991).

Whereas the laboratory studies indicated that *Bagous hydrillae* was a generalist, in the field it is able to complete its life cycle on a narrow range of hosts. The difference in host specificity between feeding, oviposition and development host-range studies completed in the laboratory and the extensive collections conducted in the field is believed to be caused by restrictions due to the terrestrial part of the life cycle. Most plants probably do not fragment in the same way that hydrilla does, so the larvae are not able to pupate in the soil and complete their life cycle.

Damage

The damage caused by the weevils is two-fold: larvae mining in the stems (Figure 75) and adults feeding around the leaf nodes (Figure 76). The adult feeding weakens the upper portions of the plant and the stems will break causing fragmentation, whereby the mat of hydrilla breaks away from the roots and usually becomes stranded on the shoreline (Center et al. 2013). Additionally, adults feeding on the leaves create distinctive holes that have been described as “pepper shot” (Balciunas and Purcell 1991). Each larva tunnels through an average of 7.5 cm of stem before emerging from the stem to pupate (Balciunas and Purcell 1991). The galleries produced by the larvae turn black (O’Brien and Askevold 1992).

Importance as a Biological Control Agent

The hydrilla stem weevil was selected as a potential biocontrol agent due to the insect’s ability to damage hydrilla in its native range in Australia. Researchers in Australia reported that the damage caused by the larvae fragmenting the hydrilla created a mowed effect, removing the top 40 inches (100 cm) (Balciunas et al. 2003, Balciunas and Purcell 1991). Host specificity testing revealed that although the weevils are opportunistic and will feed, oviposit and successfully develop on many plant species in the laboratory, in the field their requirements for pupation make them more host specific.

A release permit was granted by the U.S. Department of Agriculture Animal and Plant Inspection Service, Plant Protection Quarantine Unit (USDA APHIS PPQ), and 320,000 adult *Bagous hydrillae* were released from 1991 to 1996. During this time, 100 releases were made at 19 sites in four states: Florida, Georgia, Alabama and Texas (Center et al. 2013). Initially, it seemed like the weevils were established; at ten of the 19 sites, researchers were able to collect weevils up to a maximum duration of 4.5 years later. However, the later collections were in very low numbers, and continued efforts to find populations were unsuccessful so the project was believed to have failed (Center et al. 2013).

This belief was recently challenged when researchers found *Bagous* weevils in pitfall trap samples in Louisiana (Center et al. 2013). The samples were collected during a study to test a new trap design for sampling insects associated with aquatic plants (Parys and Johnson 2011). Although these captures were 580 km from the nearest release site, two weevils were confirmed to be the introduced *Bagous hydrillae* (Center et al. 2013). Interestingly, these weevils were collected from an area dominated by common salvinia, *Salvinia minima* Baker (Polypodiopsida: Salviniaceae), with no hydrilla presence recorded one year after the samples were collected (Center et al. 2013). Salvinia was not included in the host range tests completed prior to release of this insect in the U.S. (Buckingham and Bennett 1994).



Figure 75. Damage caused by a larva of the hydrilla stem weevil, *Bagous hydrillae*. Photograph by U.S. Army Engineer Research and Development Center.



Figure 76. Damage caused by the adult hydrilla stem weevil, *Bagous hydrillae*. The green arrow indicates the presence of the weevil adult feeding damage. Photograph by Gary Buckingham, USDA-ARS.



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BENEFICIAL/Bagous_hydrillae.
htm](http://entnemdept.ufl.edu/creatures/BENEFICIAL/Bagous_hydrillae.htm)

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the article.)

The lack of establishment of the hydrilla stem weevil is most likely due to specific requirements of the life cycle. The larvae of *Bagous hydrillae* require relatively dry conditions to pupate. In its native range in Australia, the hydrilla fragments and the mat damaged by the adults will drift to the edge of the water body. Larvae leave the damp hydrilla and pupate in the relative dryness of previously stranded hydrilla or the soil. In Florida and most of the southeastern U.S. where this insect has been released, these conditions (i.e., stranded hydrilla around water bodies) are not common and so the weevil is unable to successfully complete its life cycle (Grodowitz et al. 1995).

Monitoring and Management

Several methods can be used to monitor weevil activity. Firstly, hydrilla can be collected and the leaves examined for the characteristic damage caused by the adult weevils. Stems may be dissected and larvae also may be viewed under a light microscope. However, Berlese funnels were found to be the most effective method of extracting adults and larvae from plant material (Balciunas and Purcell 1991). Monitoring for adult weevils can be done by using ultraviolet (UV) black lights (Buckingham and Balciunas 1994). Although unintentional, *Bagous hydrillae* also were collected by using floating pitfall traps (Center et al. 2013, Parys and Johnson 2011). Management is not necessary for the hydrilla stem weevil as this insect is not a pest in the U.S.

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Featured Creatures: The Hydrilla Tuber Weevil

By Emma N.I. Weeks

SUMMARY: This article focuses on the hydrilla tuber weevil. The weevil was imported for the biological control of hydrilla but did not establish in Florida. The online version of this article is available on the UF/IFAS Entomology and Nematology department website Featured Creatures. URL: http://entnemdept.ufl.edu/creatures/BENEFICIAL/Bagous_affinis.htm

COMMON NAME: hydrilla tuber weevil (unofficial)

SCIENTIFIC NAME: *Bagous affinis* Hustache (Insecta: Coleoptera: Curculionidae)

Introduction

Bagous affinis Hustache (Figure 77) is a semi-aquatic weevil that feeds on the aquatic invasive plant *Hydrilla verticillata* (L.f.) Royle. The larvae of the weevil mine hydrilla tubers, and the adults feed on the submerged stems and leaves. The weevil was discovered during surveys for biological control agents for hydrilla in Pakistan in 1980 and was first introduced to the U.S. in Florida from India in 1987. A closely related weevil from Australia, *Bagous hydrillae* O'Brien (Insecta: Coleoptera: Curculionidae), was introduced to the U.S. in 1991.

Distribution

Bagous affinis is native to India and Pakistan. It feeds on stranded hydrilla above the waterline around water bodies in areas that have a long dry season (Buckingham and Bennett 1994).

Description

EGGS: Eggs are elongated and 0.52 mm long and 0.32 mm wide (Buckingham and Bennett 1994). The egg is white, and the chorion or outer membrane is transparent (Figure 78). The larvae can often be seen moving around in mature eggs (Buckingham and Bennett 1994). Eggs are laid in waterlogged wood, hydrilla stems, and soil.

LARVAE: There are three larval stages, and all three instars are white with brown heads (Figure 79). The first instars hatch from the eggs and appear more dorsoventrally flattened than the later instars (Buckingham and Bennett 1994). The setae are more noticeable in this stage (Buckingham and Bennett 1994). The instars can be differentiated by head capsule width, which in the first instar is 0.22 mm (Buckingham and Bennett 1994). The head capsule width in the second instar is 0.40 mm and that of the third instar is 0.66 mm (Buckingham and Bennett 1994). The first instar bores into the tubers of hydrilla, and larval development occurs within the tuber.

PUPAE: Pupae of *Bagous affinis* (Figure 80) are off white to yellow with small red-brown bristles on the head and dorsum (Buckingham and Bennett 1994). Pupae are 3.75 mm long and 1.81 mm wide (Buckingham and Bennett 1994). The pupae turn yellow as they approach pupation. One day before eclosion, adult features become apparent, the eyes and leg joints darken, and the tips of the mandibles turn red (Buckingham and Bennett 1994).



Figure 77. Adults of the hydrilla tuber weevil, *Bagous affinis* on a tuber of *Hydrilla verticillata*. Photograph by Gary Buckingham, USDA-Agricultural Research Service.



Figure 78. Egg (top) and a larva of the hydrilla tuber weevil, *Bagous affinis*. Photograph by USDA-Agricultural Research Service, Bugwood.org.



Figure 79. Larva of the hydrilla tuber weevil, *Bagous affinis* on a tuber of hydrilla, *Hydrilla verticillata*. Photograph by USDA-Agricultural Research Service.

ADULTS: Adult *Bagous affinis* are small brown weevils with light-colored mottling (Figure 81). Adults are on average 3.5 mm long and 1.5 mm wide (Buckingham and Bennett 1994). The males and females can be distinguished by the shape of the first abdominal sternite. The sternite is concave in males and convex in females (Buckingham and Bennett 1994).

Life Cycle and Biology

Life stages include the egg, three larval instars, pupa, and adult. The life cycle of *Bagous affinis* occurs in stranded plant material at the edge of water bodies when the water level recedes during dry periods (Buckingham and Bennett 1994). The adults prefer to lay eggs in waterlogged wood although they also will lay eggs in soil and into hydrilla stems (Buckingham and Bennett 1991). The female probes the substrate with her mouthparts and then inserts an egg into the probed area (Buckingham and Bennett 1994). In the hydrilla stems, eggs were laid most commonly near the leaf nodes. After 3-4 days (Buckingham and Bennett 1994), the first-instar larvae hatch and locate and mine the tubers of the hydrilla. They do not attack submersed hydrilla tubers, only those above the water level. Larval development, through the three instars, takes 14-15 days, and the larvae usually stay within the same tuber (Buckingham and Bennett 1994). The larvae will die if the tubers become submerged when the water level rises.

Pupation occurs in terrestrial habitats usually in stranded plant material or in the silt at the edge of the water body. The pupation period lasts for 5 days (Buckingham and Bennett 1994). The total time for immature development is around 22 days, and adults live for around 120-130 days on average (Buckingham and Bennett 1994). Successful development occurs between 64°F (18°C) and 90°F (32°C) (Godfrey and Anderson 1994a). *Bagous affinis* adults cannot swim, but they can remain submerged for several hours. They have been observed going underwater by clinging to hydrilla stems (Buckingham and Bennett 1994). When underwater, the weevils breathe using plastron respiration, i.e., air that is trapped close to the body by scales (Buckingham and Bennett 1994).

The larvae of a closely related species, the hydrilla stem weevil, *Bagous hydrillae*, feed on hydrilla stems. Although the female hydrilla stem weevil will lay eggs on tubers and the larvae will develop normally, the opposite is not true. The hydrilla tuber weevil larvae are not able to complete development on stem tissue and die before the prepupal stage (Wheeler and Center 2007).

Hosts

Host specificity studies were completed by Buckingham and Bennett (1998). In these tests, larvae were observed feeding on seven plant species of the 39 species in 20 families that were tested. Larvae developed on Brazilian waterweed (*Egeria densa* Planch.), dwarf arrowhead (*Sagittaria* spp.), American pondweed (*Potamogeton nodosus* Poir.), sago pondweed (*Potamogeton pectinatus* L.), Richardson's pondweed (*Potamogeton richardsonii* (A. Benn.) Rydb.), watercelery (*Vallisneria americana* Michx.), and hydrilla. However, very low levels of development occurred in all other plants compared to hydrilla.

For adults, 52 plant species in 29 families were tested. With the exception of sago pondweed, feeding was limited to plants in the Hydrocharitaceae. Plants within the family Hydrocharitaceae that were fed upon by adults included Brazilian waterweed, watercelery, and frogbit (*Limnobium spongia* (Bosc.) Rich. ex Steud.). Females laid eggs on only three out of six species, namely Brazilian waterweed, watercelery and elodea (*Elodea canadensis* Michx.).



Figure 80. Pupa of the hydrilla tuber weevil, *Bagous affinis* in a tuber of hydrilla, *Hydrilla verticillata*. Photograph by U.S. Army Engineer Research and Development Center and USDA-ARS.



Figure 81. Adult of the hydrilla tuber weevil, *Bagous affinis*. Bar equals 1 mm. Photograph by Michael Shillingburg, Arizona State University Hasbrouck Insect Collection (ASUHIS, 2013).



Figure 82. Damage caused by the larvae of the hydrilla tuber weevil, *Bagous affinis*, feeding on hydrilla tubers. Photograph by USDA-Agricultural Research Service, Bugwood.org.

Damage

Adult *Bagous affinis* feed on hydrilla leaves, stems, turions, and tubers, although stems are preferred (Buckingham and Bennett 1994). When feeding, the adults often form aggregations with all individuals feeding on the same stem or tuber (Buckingham and Bennett 1994). On the stems, adults eat the tissue around the leaf nodes and often cause the stems to break. On the tubers, adults bore into and feed on the tissue as they move into the tuber. The larvae stay within the tubers, consuming plant tissue as they develop (Figure 82). This feeding causes degradation of the tubers, so the tuber often fails to germinate once damaged.

Importance as a Biological Control Agent

The hydrilla tuber weevil was selected as a potential biocontrol agent due to the organism's ability to damage hydrilla in the native range of India and Pakistan. Baloch et al. (1980) reported that *Bagous affinis* infested nearly all of the tubers collected at one field site in Pakistan. Infested tubers were less likely to germinate than uninfested tubers (Godfrey and Anderson 1994b).

In 1987, *Bagous affinis* was released in Florida and was the first insect species ever released for hydrilla control (Grodowitz et al. 1995). Releases continued in Florida until 1988. In July 1990, there was no establishment of the weevil at the release sites (Buckingham and Bennett 1994). The most likely reason for the lack of establishment of the hydrilla tuber weevil is the specific requirements of the life cycle. The larvae of *Bagous affinis* require relatively dry conditions for larval development and pupation. In the native range of India and Pakistan, the weevil completes its whole life cycle in stranded hydrilla at the edge of water bodies.

In Florida and most of the southeastern U.S., where this insect has been released, these conditions (i.e., stranded hydrilla around water bodies) are not common and so the weevil is unable to successfully complete its lifecycle. The only evidence of field colonization, albeit temporary, was during a drawdown in a reservoir in north-central Florida (Buckingham and Bennett 1991). A drawdown is a method of hydrilla control used in water bodies with water level control structures. The water level is decreased so that the hydrilla dries out and dies. Unfortunately, the insects were killed when that water level returned to normal levels.

In 1992, it was realized that, although the necessary conditions for establishment of this species were not present in the southeastern U.S., *Bagous affinis* may be able to establish in other areas, specifically California. Releases were made to determine if *Bagous affinis* would establish and successfully overwinter in a river and a pond in northern California (Godfrey et al. 1994). No establishment occurred at the river site (Chowchilla River), but it was later discovered that not many tubers were present (Godfrey et al. 1994). At the pond site (Calaveras County), the weevils overwintered in cages over two years (Godfrey et al. 1994). When the eggs were released directly into the pond (i.e., not in cages), temporary establishment occurred over the summer with tuber damage visible on sentinel tubers (Godfrey et al. 1994). Although no adults were caught in the spring of the following year, tubers from the sentinel traps showed characteristic weevil damage (Godfrey et al. 1994).

In 1994, *Bagous affinis* was released at Choke Canyon Reservoir in Texas (Grodowitz et al. 1995). It was believed that this site would be conducive to weevil development as the water level could be altered and stranded tubers were visible at a rate of 90 tubers per m² (Grodowitz et al. 1995). However, when the soil and tubers were examined three months later, no individuals were found and no damage was recorded (Grodowitz et al. 1995).

Monitoring and Management

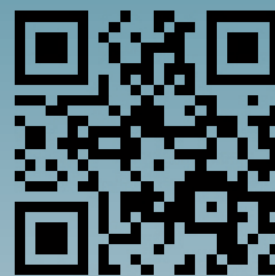
Several methods can be used to monitor weevil activity including plant dissection and black lights. Firstly, hydrilla can be collected and the leaves examined for the characteristic damage caused by the adult weevils. Tubers may be dissected and larvae may be visualized under a light microscope. Monitoring for adult weevils can be done using ultraviolet (UV) lights (Buckingham and Bennett 1994). Management is not necessary for the hydrilla tuber weevil as it is not a pest in the U.S.

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Featured Creatures: The Waterlily Leafcutter

By Dale H. Habeck, James P. Cuda, and Emma N.I. Weeks

SUMMARY: This article focuses on the waterlily leafcutter. This moth is a potential biological control agent of hygrophila, *Hygrophila polysperma* (Roxb.) T. Anderson (Polemoniales: Acanthaceae). However, the moth is a generalist, and the larvae have been observed frequently to eat hydrilla. The online version of this article is available on the UF/IFAS Entomology and Nematology department website Featured Creatures. URL: http://entnemdept.ufl.edu/creatures/beneficial/leps/waterlily_leafcutter.htm

COMMON NAME: waterlily leafcutter

SCIENTIFIC NAME: *Elophila oblitalis* (Walker) (Insecta: Lepidoptera: Crambidae: Acentropinae)

Introduction

Hygrophila polysperma (Roxb.) T. Anderson (Polemoniales: Acanthaceae) is a rooted submersed or emersed aquatic plant in shallow water areas and saturated shorelines throughout Florida. This invasive aquatic plant also is known as hygrophila, hygro, East Indian hygro, green hygro, Miramar weed, oriental ludwigia, and Indian swampweed (hereafter referred to as hygrophila).

Hygrophila is a federal listed noxious weed (USDA 1983), a Florida state listed Category II prohibited plant (FLDEP 1993), and a Florida Exotic Pest Plant Council Category I invasive species (FLEPPC 2007). The submersed growth habit displaces native vegetation in many canals and drainage ditches in south Florida. The plant forms dense stands that occupy the entire water column, clogging irrigation and flood-control systems (Schmitz and Nall 1984, Sutton 1995) and interfering with navigation (Woolfe 1995). Hygrophila also creates problems as an emergent plant in some shoreline areas, including rice fields (Kromholz 1996).

In October 2007, we received a report from researchers at the UF/IFAS Center for Aquatic and Invasive Plants of an insect attacking hygrophila. Samples of the insect were collected and it was identified as the waterlily leafcutter *Elophila oblitalis* (Walker). Of the more than twenty Acentropinae species occurring in Florida, *Elophila oblitalis* (Walker) is the most common. Although its common name implies that it is a pest of waterlilies, it actually has a wide host range. Most of the damage caused by the larvae usually is superficial and rarely endangers the plant, but the damage observed on the hygrophila plants was severe (Figures 83 and 84).

In addition to the invasive aquatic weed hygrophila, the waterlily leafcutter also feeds on another invasive plant, hydrilla, *Hydrilla verticillata* L.f. Royle. Numbers of *Elophila oblitalis* collected from hydrilla from field sites in Florida and Louisiana were similar to the numbers of the hydrilla leafcutter moth, *Parapoynx diminutalis* Snellen that were collected (Balciunas and Minno 1985).



Figure 83. *Hygrophila* showing feeding damage caused by the waterlily leafcutter, *Elophila oblitalis* (Walker). Photograph by James Cuda, University of Florida.



Figure 84. Larva of the waterlily leafcutter, *Elophila oblitalis* (Walker), attacking *hygrophila*. Photograph by James Cuda, University of Florida.



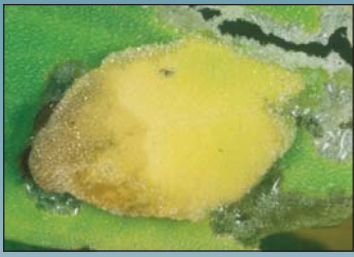


Figure 85. Waterlily leafcutter, *Elophila oblitalis* (Walker), leaf case. Photograph by Lyle Buss, University of Florida.



Figure 86. Larva of the waterlily leafcutter, *Elophila oblitalis* (Walker), with opened leaf case. Photograph by Lyle Buss, University of Florida.



Figure 87. Pupa of the waterlily leafcutter, *Elophila oblitalis* (Walker). Photograph by Stephen Luk, Richmond Hill, Ontario, Canada.

Synonymy

According to Dyar (1906) and Zimmerman (1958) the following synonyms have been used for *Elophila oblitalis*:

- Synclita oblitalis* Walker, 1859
- Synclita proprialis* Fernald, 1859
- Isopteryx oblitalis* Walker, 1859
- Parapoynx obscuralis* Walker, 1859
- Parapoynx obscuralis* Möschler, 1972
- Hydrocampa proprialis* Fernald, 1888
- Hydrocampa oblitalis* Fernald, 1891
- Nymphula oblitalis* Hampson, 1897

Distribution

This common moth occurs throughout Florida, westward to Texas and northward to western Nova Scotia and southern Manitoba (Munroe 1972). It also has been introduced into Hawaii (Williams 1944), England (Shaffer 1968), and British Columbia (Munroe 1972).

Description

EGGS: The eggs are whitish in color, and appear domelike (oval and flattened). The flattened side is glued to the leaf and the domed side has wrinkles down the length of the egg (Dyar 1906). The eggs are 0.6 mm in length and 0.4 mm wide (Dyar 1906). They are deposited singly or in overlapping, ribbon-like masses near the edges of submersed leaf surfaces.

LARVAE: Most members of the crambid subfamily Acentropinae have aquatic larvae with tracheal gills. However, the larva of this moth lacks gills and lives between two pieces of leaf (leaf case) that it cuts from its host plant (Figure 85 and 86).

The epidermis (skin) of the larvae is covered with minute papillae (bumps). The body is creamy-white, but increasingly brownish from abdominal segment four forward to the prothorax. The prothoracic coxae (proximal leg segments) are touching while the mesothoracic coxae are nearly touching. The head is yellowish-brown with a faint brown genal (cheek) stripe. The prothoracic spiracle (respiratory opening) is vestigial (non-functioning), and the while spiracles on abdominal segments three and four are distinctly larger than others. The crochets (gripping hooks) are arranged in two biordinal (sometimes partially triordinal) transverse bands, with the anterior band distinctly larger than the posterior band.

PUPAE: The pupae are pale yellow and the wings and head appear darker (Figure 87) (Dyar 1906). The head has two distinct black spine-like hairs. The spiracles on abdominal segments 2-4 are large, round, elevated and red brown in color (Dyar 1906). The anterior spiracles are much smaller. The pupae are found within silk cocoons within the leaf cases formed by the larvae.

ADULTS: Adults are sexually dimorphic and readily distinguishable (Figures 88 and 89). Females have a 15 to 19 mm wingspan, and the female's wings are paler in color appearing grayish-brown with orange-brown markings. The wingspan of the male is only about 11 to 13 mm, and the male's wings are grayish-brown interspersed with brownish and white markings.

Life Cycle and Biology

No information is available about the development times of this species. The female moth lays her eggs on the exposed edges of submersed aquatic plants (Gill et al. 2008). Upon hatching, the larvae enclose themselves inside cut leaf pieces. The leaves are webbed together with silk. Cases made by young larvae are water-filled and oxygen uptake occurs cutaneously (presumably via the epidermal papillae) whereas cases of older larvae are air filled. The cases of young larvae remain attached to the leaf from which they were made. Older larvae detach the case from the leaf and are free-floating.

Larvae abandon smaller cases as they mature and construct larger cases from new leaves. The case may consist of two entire leaves, parts of leaves, or of parts of many plants tied together with silk. The larvae extend out of the case to feed on plant material, but usually the body remains in the case. Prior to pupation, larvae attach their cases to petioles or leaf blades of their host plants above or below the water surface, and spin a silk cocoon inside their leaf cases.

Hosts

Elophila oblitalis has a wide host range and is known to feed on nearly 60 plant species (see Table on page 114).

Damage

Elophila oblitalis has a wide host range and is known to feed on waterlilies and other ornamental pond plants as well as the invasive aquatic weeds, hygrophylla and hydrilla. The larvae are the stage that feeds on the plant and causes damage to the plant tissue. In addition to feeding, the larvae cut the leaves to prepare a leaf case for shelter. As the larvae develop, they cut new, progressively larger leaf cases. This action in itself can provide quite significant damage to the infested plant (Nachtrieb et al 2007).

In a field study, to compare the effect of herbivory on different aquatic plants, *Elophila oblitalis* was one of the three species that caused the most damage (Nachtrieb et al 2007). When feeding, the larvae remove chunks from the leaves, usually feeding on the basal or middle portions (Balciunas and Minno 1985). This feeding often causes the leaves to break away from the stem. If the population density is high and plant material becomes more scarce, the larvae will begin to feed on the stems, which can cause the entire plant to fragment (Balciunas and Minno 1985).

Due to its broad host range, this insect frequently is a pest in aquatic plant nurseries, especially on waterlilies, *Nymphaea* spp. In the nursery setting, this insect can cause economic losses as the larval feeding makes the plants unattractive to customers. Extensive feeding may even lead to reduced plant health and death (Gill et al. 2008).



Figure 88. Adult female waterlily leafcutter, *Elophila oblitalis* (Walker). Photograph by J. Lotz, Division of Plant Industry.



Figure 89. Adult male waterlily leafcutter, *Elophila oblitalis* (Walker). Wingspan of this specimen is 11 mm. Photograph by Lyle Buss, University of Florida.

This table shows the host range of the waterlily leafcutter. Plants are arranged by families and genera that are known to be hosts for *Elophila oblitalis*.

Family	Genera	Number of species
Acanthaceae	<i>Hygrophila</i>	1
	<i>Nomophila</i>	1
	<i>Synema</i>	1
Alismataceae	<i>Enchinodorus</i>	3
	<i>Sagittaria</i>	1
Amaranthaceae	<i>Amaranthus</i>	1
Apiaceae	<i>Hydrocotyle</i>	3
Aponogetonaceae	<i>Aponogeton</i>	3
Araceae	<i>Orontium</i>	1
	<i>Pistia</i>	1
Brassicaceae	<i>Cardamine</i>	1
	<i>Nasturtium</i>	1
Cyperaceae	<i>Eleocharis</i>	1
Gentianaceae	<i>Nymphoides</i>	2
Haloragaceae	<i>Myriophyllum</i>	2
Hydrocharitaceae	<i>Egeria</i>	1
	<i>Elodea</i>	1
	<i>Hydrilla</i>	1
	<i>Limnobium</i>	1
Lemnaceae	<i>Lemna</i>	1
	<i>Spirodela</i>	1
Lythraceae	<i>Rotala</i>	1
Marsileaceae	<i>Marsilea</i>	1
Nymphaeaceae	<i>Brasenia</i>	1
	<i>Nelumbo</i>	1
	<i>Nuphar</i>	1
Onagraceae	<i>Nymphaea</i>	7
	<i>Ludwigia</i>	2
Poaceae	<i>Hydrochloa</i>	1
Polygonaceae	<i>Polygonum</i>	3
Pontederiaceae	<i>Eichhornia</i>	1
	<i>Pontederia</i>	1
Potamogetonaceae	<i>Potamogeton</i>	3
Salicaceae	<i>Salix</i>	1
Salviniaceae	<i>Azolla</i>	1
	<i>Salvinia</i>	1
Scrophulariaceae	<i>Ambulia</i>	1
	<i>Bacopa</i>	1
	<i>Lindernia</i>	1
	<i>Micranthemum</i>	1

Importance as a Biological Control Agent

In addition to having a pest status in aquatic nurseries, due to its wide host range, *Elophila oblitalis* also plays a minor role in biological control, as it feeds on invasive species, such as hydrilla and hygrophila. As a native species this type of biological control is known as natural regulation. However, due to the extensive host range of this species it would not be advisable to attempt to increase wild numbers through mass releases or conservation as they would likely feed non-specifically on other desirable plants as well as the weeds.

Monitoring and Management

To monitor for the waterlily leafcutter, observe leaves for the characteristic holes created by this insect (Gill et al. 2008). The adults can be trapped by UV black lights and the larvae can be extracted from the plant material by handpicking or using a Berlese funnel.

Elophila oblitalis is a pest of greenhouses and may require control in aquatic plant nurseries. As with other aquatic moth pests, *Bacillus thuringiensis* subspecies *kurkstaki* would likely provide control with little or no adverse effects to other aquatic organisms. In support of this hypothesis, the closely related organism *Bacillus thuringiensis* subspecies *israelensis* was found to cause significant mortality to the waterlily leafcutter (Haag and Buckingham 1991).

Related Species

Three other species of *Elophila* occur in the United States with one, *Elophila tinealis* Munroe, in Florida. The adult of *Elophila tinealis* is much smaller than that of the waterlily leafcutter and has longer, narrower and darker wings. The larvae of *Elophila tinealis* are not well known, but seem to feed on and most often make their cases of duckweed, *Lemna* sp.

The larvae of *Elophila gyralis* (Hulst) and *Elophila icciusalis* (Walker) are similar to those of the waterlily leafcutter, but the anterior and posterior transverse bands of crochets (the gripping hooks on the prolegs) are the same size. *Elophila gyralis* and *Elophila icciusalis* adults are more brightly colored than *Elophila tinealis* and *Elophila oblitalis* and are yellowish-orange and white or brownish in color. Although *Elophila gyralis* and *Elophila icciusalis* larvae may make portable cases, they usually cut only one leaf piece and attach it to a whole leaf and live between the two layers.

Acknowledgements

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Featured Creatures: The Asian Grass Carp

By Emma N.I. Weeks and Jeffrey E. Hill

SUMMARY: This article focuses on the Asian grass carp. The carp is a generalist herbivore of aquatic plants. It was imported for biological control of hydrilla and is used extensively for aquatic plant management throughout the U.S. The online version of this article is available on the UF/IFAS Entomology and Nematology department website Featured Creatures. URL: http://entomology.ifas.ufl.edu/creatures/BENEFICIAL/MISC/Ctenopharyngodon_idella.htm

COMMON NAME: grass carp, the white amur

SCIENTIFIC NAME: *Ctenopharyngodon idella* Cuvier and Valenciennes
(Actinopterygii: Cyprinidae: Squaliobarbinae)

Introduction

The grass carp, *Ctenopharyngodon idella* Cuvier and Valenciennes, was imported to the U.S. in 1963 as a biological control agent for hydrilla (*Hydrilla verticillata* (L.f.) Royle) and other aquatic plants. Efficacy experiments were conducted in Florida in the 1970s by the United States Department of Agriculture and the University of Florida. Use of the fish was limited from 1970 until 1984 due to tight regulations surrounding concerns of escape and reproduction, and the potential impacts that colonization of the fish could have on native flora and fauna. These concerns led to research that developed a non-reproductive fish, which was equally effective in controlling hydrilla.

Sterile fish were developed by subjecting eggs to stress, such as heat stress (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 virtually sterile, this does not affect their aquatic plant herbivory. Concern over the success rate of the sterilization technique led to screening for diploid individuals by measuring the diameter of cell nuclei, as triploid cells have larger nuclei. In the warm waters of Florida, with abundant food, grass carp grow quickly at around 2 lbs/month or 0.91 kg/month and may achieve weights of 97 lbs (44 kg) (Sutton et al. 2012). Younger fish and female fish grow faster than older or male fish.

Grass carp are the most effective biological control tool that has been identified for hydrilla. Additionally, although conversion of plant material to protein by the grass carp is not highly effective, it is still the best use for hydrilla. Every 1 lb (0.45 kg) increase in fish weight requires 5-6 lbs (2.3-2.7 kg) of dry hydrilla (Sutton et al. 2012), which—considering hydrilla is 95% water—is a great deal of live plant material.

Synonymy

According to Shireman and Smith (1983) the following synonyms have been used for *Ctenopharyngodon idella*:

Leuciscus idella Cuvier and Valenciennes 1844

Leuciscus tschiliensis Basilewsky 1855

Ctenopharyngodon laticeps Steindachner 1866

Sarcocheilichthys teretiusculus Kner 1867

Ctenopharyngodon idellus Günther 1868

Pristiodon siemionovi Dybovskii 1877



Distribution

The grass carp is native to rivers that feed into the Pacific Ocean in eastern Russia and China, but it has been introduced to 70 countries including the U.S., Taiwan, Japan, Mexico, India, Malaysia, and several European countries. In the U.S., grass carp are so effective for weed control that they are used nationwide. In 2009, the use of grass carp was recorded in 45 states, all states except Alaska, Maine, Montana, Rhode Island, and Vermont. Within the native range of the grass carp, the natural habitat includes low-gradient, large turbid rivers and associated lakes. Grass carp are highly temperature tolerant, and their native range includes both cold and warm water environments. Early release of diploid fish led to reproductive populations in several U.S. drainage systems, including the Mississippi River and major tributaries.

Within the U.S., the distribution in water bodies is widespread, particularly in the Mississippi River basin and southeastern states (Figure 90). In Figure 90, distribution of the grass carp is classified by drainage system at two scales, fine and medium. Medium scale or HUC 6 is known as a basin and is on average 10,600 square miles in area. Fine scale or HUC 8 is known as a sub-basin and is on average 700 square miles in area. Occurrence of grass carp within a basin or sub-basin results in highlighting the entire drainage system. Drainages with reproductive, established populations are much less prevalent than suggested by the overall distribution of stocked and reported grass carp shown in Figure 90, many of which are non-reproductive triploids. Established populations occur in the Mississippi River basin and some drainages of eastern Texas.

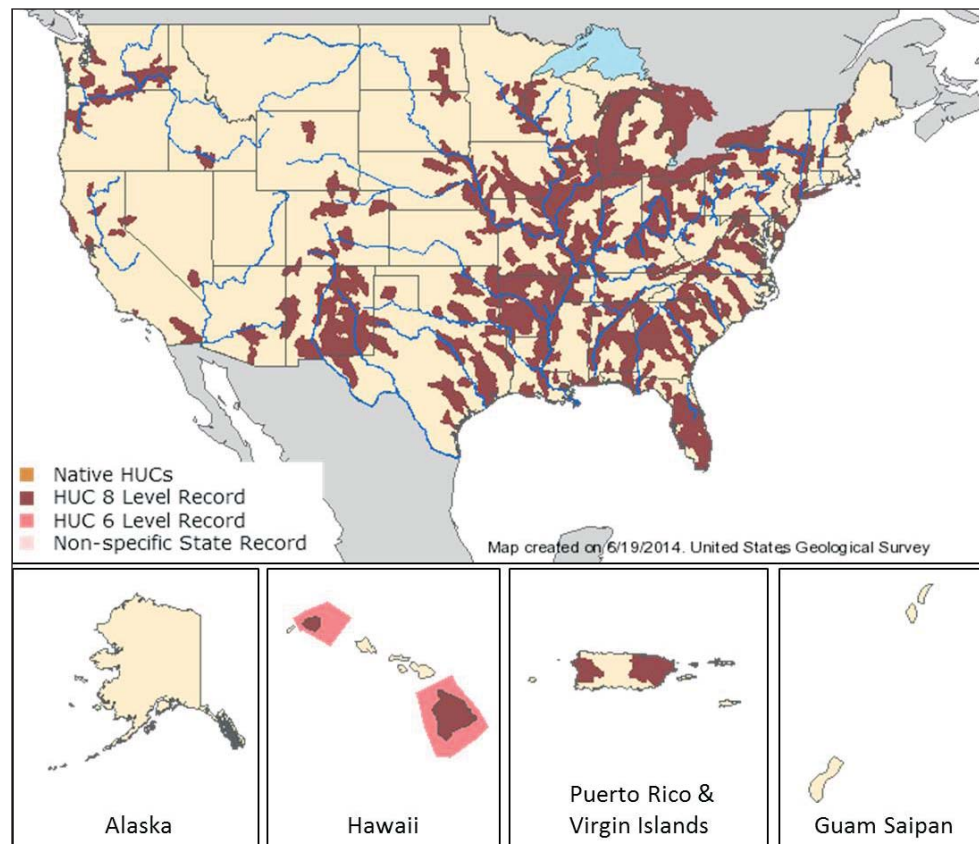


Figure 90. Distribution of grass carp, *Ctenopharyngodon idella* Val., in the United States as reported in the Nonindigenous Aquatic Species database at the U.S. Geological Survey (USGS). Map reproduced with permission from NAS.

Description

EGGS: Unfertilized eggs are 1.2 - 1.3 mm in diameter and have a yolk surrounded by a double-layered membrane (Shireman and Smith 1983; Figure 91). The outer layer is adhesive until fertilization (Shireman and Smith 1983). Fertilized eggs are 3.8 - 4.0 mm in diameter, and the yolk is separated from the membrane by water that is absorbed (Shireman and Smith 1983). Spawn containing eggs can be grayish-blue to bright orange (Shireman and Smith 1983).

PROTOLARVAE (DAYS 1-3): Protolarvae hatch from the eggs at 5.0 - 5.5 mm in length (Figure 92). At this stage, they are transparent and completely without pigment. Within three days, they grow to 7.4 - 7.5 mm and develop useable gills. At this stage, the eyes become pigmented with gold irises, and the head and dorsum are green/yellow. During this time, protolarvae also begin to swim. Although protolarvae still are feeding mainly from the yolk sac, from day 2, the larvae will start to eat algae.

MESOLARVAE (DAYS 4-20): By day 4, the larvae are 7.5 - 8.0 mm with a functional swim bladder and gills (Figure 93). The larvae become more motile and more pigmented every day. By day 20, the mesolarvae are 11.5 - 18.6 mm, and the fins have formed. The larvae are highly pigmented with a brown/yellow dorsum fading to white at the belly. As the yolk sac is quickly depleting, the larvae start to feed from the environment on algae and zooplankton, and by day 5 feed almost exclusively on zooplankton.

FRY (DAYS 20-30): Fry are 1.5 - 2.3 cm with well-developed fins and scales (Figure 94a). The teeth have formed, and the jaw has set. The swim bladder and the intestine resemble those of an adult. Fry feed on zooplankton and aquatic insect larvae. At 2 cm in length, the fry begin to eat aquatic plants.

FINGERLINGS (DAYS 45-60): Fingerlings are 3.7 - 6.7 cm in length and resemble small adults (Figure 94b). By day 50, the scales are complete, and at approximately day 55 and 6.7 cm in length, the fingerling is identical to an adult. Fingerlings can eat animal food (e.g., insects and zooplankton), but by 5.5 cm in length are eating mainly plants.

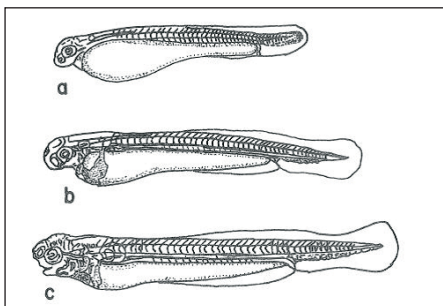


Figure 92. Protolarval development of grass carp, *Ctenopharyngodon idella* Val. a. 1 day, b. 2 days, and c. 3 days. Extracted from Shireman and Smith (1983) and used with permission from the Food and Agriculture Organization of the United Nations.

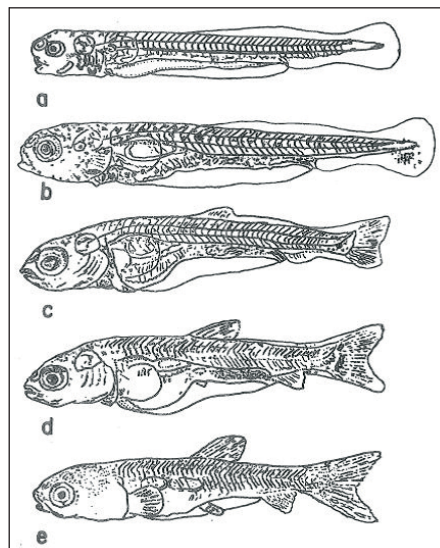


Figure 93. Mesolarval development of grass carp, *Ctenopharyngodon idella* Val. a. 4.5 days, b. 7 days, c and d. 9-18 days, and e. 20 days. Extracted from Shireman and Smith (1983) and used with permission from the Food and Agriculture Organization of the United Nations.

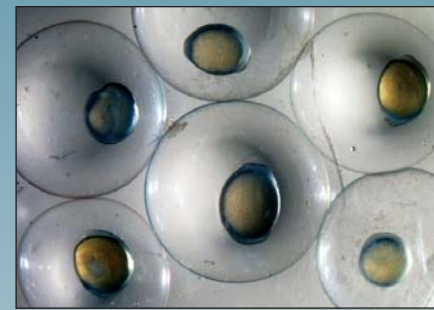


Figure 91. Eggs of carp (*Actinopterygii: Cyprinidae*). Photograph by Reuben Goforth, Purdue University.

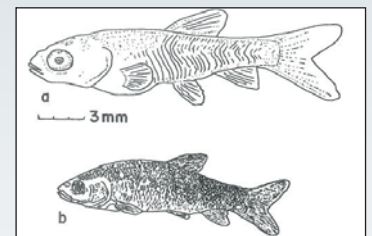


Figure 94. Postlarval development of grass carp, *Ctenopharyngodon idella* Val. a. fry and b. fingerling. Bar equals 3 mm. Extracted from Shireman and Smith (1983) and used with permission from the Food and Agriculture Organization of the United Nations.



Figure 95. Juvenile grass carp, *Ctenopharyngodon idella* Val. Photograph by Jeffrey Hill, University of Florida.



Figure 96. Adult grass carp, *Ctenopharyngodon idella* Val. Photograph by Jeffrey Hill, University of Florida.

JUVENILES (1-9 YEARS): Juveniles continue to grow and develop, but they already look identical to adults (Figure 95). The body of a juvenile or adult grass carp is torpedo shaped. The mouth angles downwards and the lips are firm and lacking barbells (i.e., fleshy whiskers). The body is dark olive in color, with brown to yellow shading on the sides and a white underside. The scales are large and outlined in brown, and the complete lateral line has 40 to 42 scales. Compared to other cyprinids, the anal fin is relatively close to the tail fin. Juveniles can feed on animal food (e.g., insects and zooplankton), but like adults, prefer to feed on plants. As the fish get larger and older, they feed on tougher plants of greater variety.

ADULTS: The maximum length of a grass carp is 4.6 ft (1.4 m), and the maximum weight is 97 lbs (44 kg). Adults look identical to juveniles (Figure 96). Adult grass carp prefer to eat hydrilla compared to all other aquatic plants.

Life Cycle and Biology

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, as the fish cannot reproduce in confined water bodies. The status of introduced grass carp populations is often difficult to determine because stocked individuals live such a long time and frequently there is little monitoring for successful recruitment. Of all the countries where the fish were introduced, they have established primarily in a few countries in Asia and Europe (Shireman and Smith 1983; Froese and Pauly 2014). However, there have been reports of several other sites having breeding populations including the Atchafalaya, Mississippi (and major tributaries), and the Trinity rivers in the U.S. (Shireman and Smith 1983, Nico et al. 2014).

In native areas, adult grass carp spawn in long fast-moving rivers at temperatures of 68-86°F (20-30°C). Spawning is triggered by increases in flow rate and temperature. Spawning generally occurs at the surface and is usually promiscuous, involving many males to each female (Shireman and Smith 1983). Fertilization occurs externally, and the semi-buoyant eggs then develop in the water column and may drift 30-100 miles (50-180 km) before hatching (Shireman and Smith 1983). Each female lays 500,000 eggs per brood on average, and fecundity increases with age (Shireman and Smith 1983). However, most eggs are lost to suffocation, disease, or predation (Shireman and Smith 1983). If the water temperature surrounding the eggs drops below 64°F or 18°C, the hatch rate and survival of larvae will be low (Shireman and Smith 1983).

Larvae have a characteristic movement that involves alternating between swimming and sinking. These larvae migrate from fast-moving rivers into lakes that act as nurseries for the juvenile fish. As juveniles, they migrate up or down stream and spend the winter in deep holes in the river bed (Shireman and Smith 1983). Juvenile grass carp feed on small invertebrates but shift to a plant-based diet by the time they reach 2 inches (5 cm) in length (Colle 2009). Female grass carp mature at 23-26 inches (58-67 cm) and males approximately one year earlier at 20-24 inches (51-60 cm). The average life of a grass carp is from 5 to 9 years. However, a grass carp may live for 20 years or more (Sutton et al. 2012).

Outside of most native areas, and for the cultivation of grass carp in the U.S. for aquatic plant management, fertilization is completed artificially. Sexually mature male and female fish are injected with hormones to promote ovulation and sperm production (Shireman and Smith 1983). Sperm, which are collected from the males, and eggs from the females are mixed and incubated with aeration to maintain movement of the eggs as they would experience in a fast-moving river.

Hosts

The grass carp is a grazer, feeding on vegetation mostly near the surface and in shallower waters. The new growth of submersed plants is preferred. Host preference is dependent on fish size, with small fish preferring musk grass (*Chara* spp.) and large fish preferring hydrilla (Sutton et al. 2012). However, the grass carp is a generalist, and in the absence of the preferred host 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.

The five most-preferred species in order of preference are hydrilla, musk grass, pondweeds (*Potamogeton* spp.), southern naiad (*Najas guadalupensis* [Spreng] Magnus), and Brazilian elodea (*Egeria densa* Planch Anderson) (Sutton et al. 2012). Grass carp are not a good control method for filamentous algae, Eurasian milfoil (*Myriophyllum spicatum* L.), spatterdock (*Nuphar advena* Aiton), fragrant waterlily (*Nymphaea odorata* Aiton), sedge (*Cladium* spp.), cattail (*Typha* spp.), or other large aquatic plants (Colle 2009).

Damage

Grass carp lack teeth in their jaws but have comb-like teeth on their pharyngeal arches (located in the throat) that enable them to grind vegetation. In fact, their scientific name means “distinctive comb pharyngeal teeth.” Small fish will eat only the leaves, but as they increase in size, they will eat both leaves and stems (Edwards 1974). As adults, they consume large amounts of plant material, preferentially hydrilla. In suitably warm water (68°F or 20°C), an adult grass carp will consume its body weight in hydrilla every day (Edwards 1974). Although adult grass carp consume a lot of plant material, the conversion to animal protein is limited. For a 1 lb (0.45 kg) increase in fish weight, the fish must eat the equivalent of 5-6 lbs (2.3-2.7 kg) of dry hydrilla (Sutton et al. 2012).

To ensure that hydrilla consumption by the fish exceeds the growth rate of the plant, several factors need to be considered, including age and sex of the fish. Depending upon these factors and the type, abundance and location of the plants within the water body, a stocking density can be determined. A study that investigated the effect of stocking rates on the ecosystem in 38 lakes in Florida found that 25 to 30 grass carp per hectare vegetation was the rate that produced the best control while leaving some less palatable species (Hanlon et al. 2000).

In the study, this was equivalent to 10 to 15 grass carp per hectare of lake area (Hanlon et al. 2000). Of the 38 lakes, 27 had a hydrilla problem (Hanlon et al. 2000). Stocking rates greater than 30 grass carp per hectare vegetation resulted in complete removal of all vegetation and rates of less than 25 grass carp per hectare vegetation resulted in insufficient control of the target plant (Hanlon et al. 2000). The Florida Fish and Wildlife Conservation Commission typically recommend stocking 7.5 to 30 fish per hectare of lake area (3 to 10 fish per acre).

An ecosystem that has been stocked with grass carp will change in several ways if the aquatic vegetation is eliminated. Phytoplankton (small floating aquatic plants) will increase and cause a decrease in water clarity (Colle 2009). Fish species that are reliant on vegetation (e.g., chain pickerel, bluespotted sunfish, and golden topminnow) will decline and may be eliminated from the ecosystem, and species that feed on phytoplankton (e.g., gizzard shad and threadfin shad) will increase in number. This species composition change has occurred in several lakes in Florida that were stocked with grass carp (Colle and Shireman 1994).

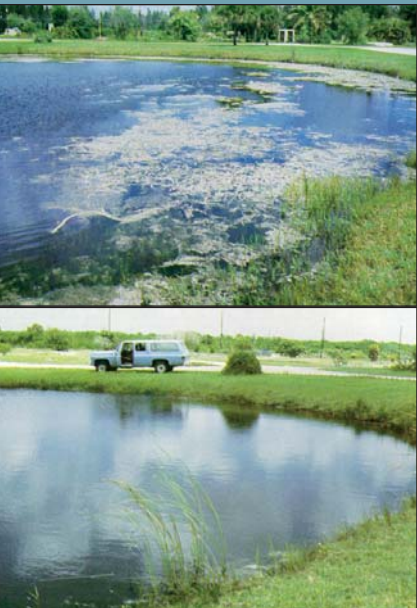


Figure 97. A pond in southeast Florida before (top) and one year after (bottom) stocking with grass carp at 40 grass carp per acre. Photographs by David Sutton, University of Florida

Importance as a Biological Control Agent

Several studies have demonstrated the effectiveness of grass carp for aquatic plant management (Figure 97). In two lakes in Florida, hydrilla infestations were eliminated in 4-5 years (Colle and Shireman 1994). In five other lakes in Florida, submersed aquatic plants were removed successfully in 1970 and remained controlled for at least 20 years (Colle and Shireman 1994).

An integrated program utilizing grass carp will be more cost effective than herbicide treatments alone. In 1994, a study estimated that, over a 9-year management program (1986 - 1994), the use of grass carp saved \$200,000 (Jaggers 1994). The Florida Fish and Wildlife Conservation Commission state on their website that grass carp may cost \$15 to \$150 per acre depending on price and stocking rate, herbicides can cost \$100 to \$500 per acre, and mechanical control around \$1,000 per acre. Additionally, while grass carp will continue to provide control, both chemical and mechanical control will need to be continuously implemented.

When introduction of a biological control agent is considered, the first condition that needs to be met is usually host specificity. Although large adult grass carp prefer hydrilla, younger smaller individuals prefer other plants. Furthermore, when hydrilla has been removed from the lake, the carp will eat other less-preferred plants. Therefore, it is important that lakes are not overstocked because the fish are difficult to remove once introduced.

Grass carp must only be stocked into closed water bodies. In open water bodies, any canals, channels or streams leading into other areas must be blocked with barriers to prevent fish escape. The barriers need to have a fine enough mesh to prevent the smallest fish from swimming through and must be high enough so that the fish cannot jump over.

Small grass carp may be lost to predation by birds, snakes, and other species of fish. In particular, largemouth bass will consume grass carp smaller than 18 inches (45 cm). In water bodies with largemouth bass it is recommended to stock fish larger than 12 inches (30 cm) or 1 lb (0.45 kg).

Every state has different regulations for the use of grass carp. Florida does not permit diploid grass carp, but some states such as Alabama allow diploid fish. Florida permits the release of triploid grass carp, but some states do not allow triploids (e.g., Maryland), and some states such as Michigan have banned the release of any grass carp. Florida requires that the released fish are certified triploid and that a permit is obtained for use, possession and removal of grass carp. Permits can be obtained from the Florida Fish and Wildlife Conservation Commission.

Monitoring and Management

Grass carp monitoring could be completed by netting or electrofishing along transects or by using hydroacoustics (Baerwaldt et al. 2013). Hydroacoustic techniques are non-invasive but do not identify fish to species. However, grass carp are rarely monitored once released.

When stocking grass carp, consider that they may eventually need to be removed once control of the aquatic weeds have been achieved. Removal is not easy, without killing all fish in the water body, and requires a permit. Several methods have been tested without much success - particularly in large water bodies - including netting, electrofishing, and rotenone treatments (Colle and Shireman 1994). Removal is usually a slow process through predation, fishing, and natural mortality. Fishing can be particularly effective in small systems.

Acknowledgements

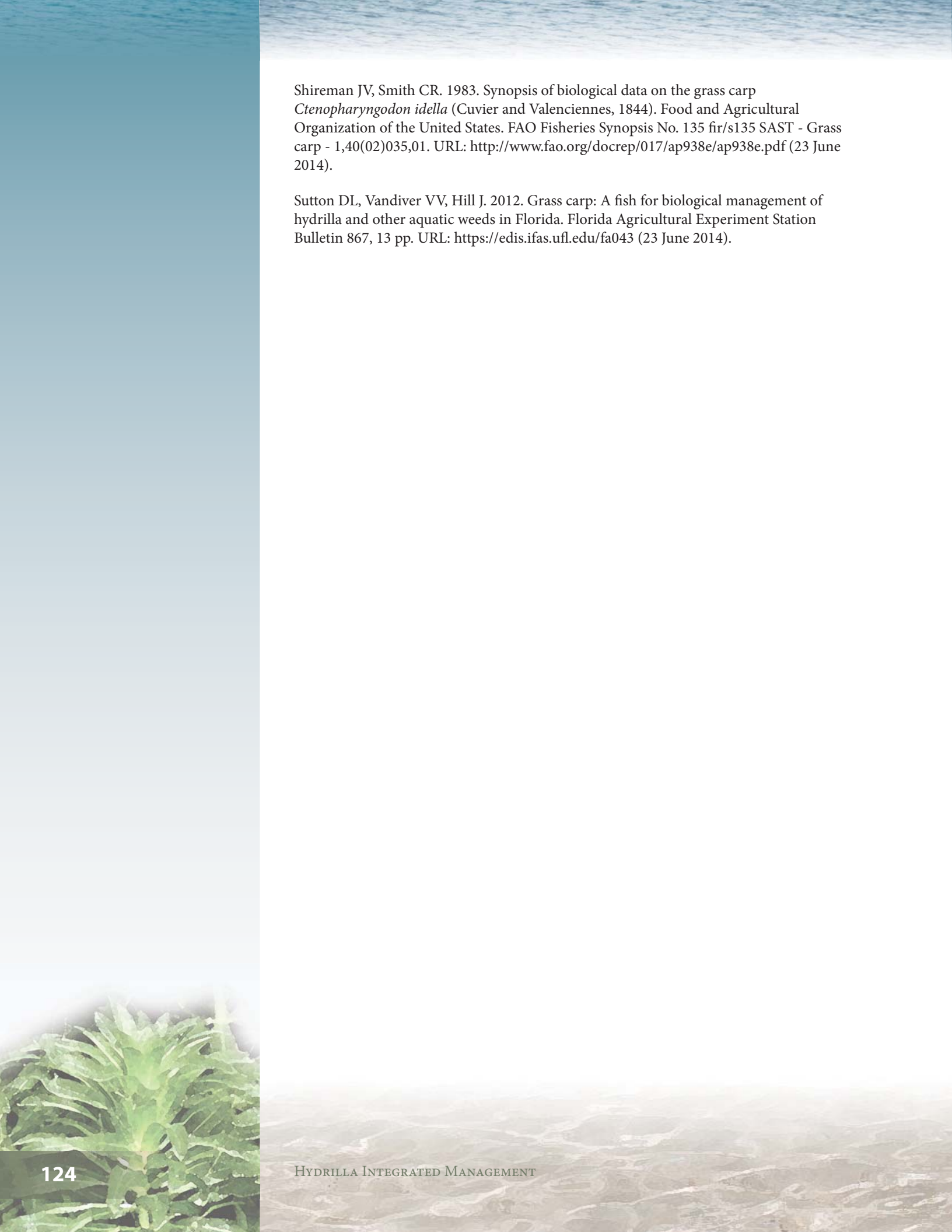
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Chapter 7

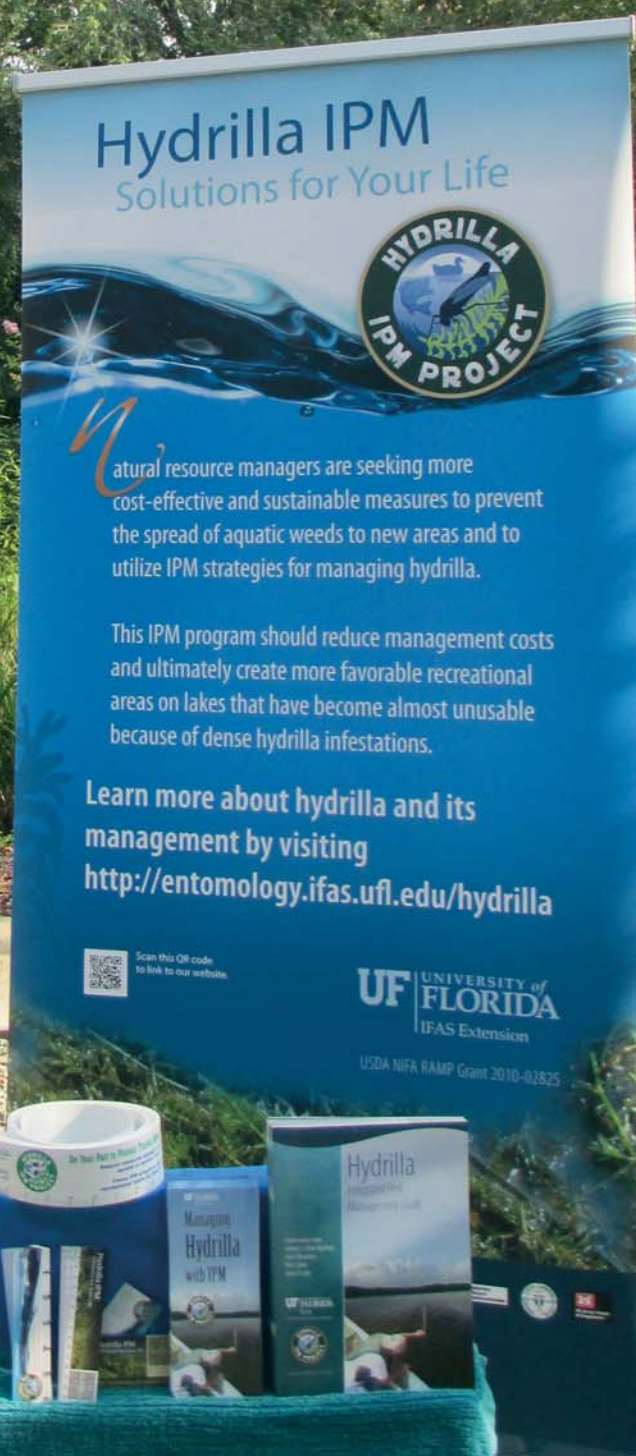


Supplementary Information

By Emma N.I. Weeks and Verena-U. Lietze

CONTENTS

Contact Information	126
Web Links to Hydrilla Fact Sheets	129
FAQs and Stakeholder Feedback	131
Spread the Word	134
Glossary	135





Find the contact information for your local UF/IFAS Extension office online. URL: <http://solutionsforyourlife.ufl.edu/map> (Scan the QR code to connect to the website.)

This chapter will provide you with useful contact information should you encounter hydrilla in your environment. It also includes links to fact sheets and a section with frequently asked questions and answers that can help you convey information to your clientele. We hope you will be inspired by our suggestions how to spread the word about hydrilla in your area. Last but not least, a short glossary explains terms that we used throughout this guide.

Contacts for Plant Identification and Management Advice

Before anybody can take steps towards weed control, they need to verify that the aquatic plant that is causing a problem is an invasive species. Expert help to identify aquatic plant species is available in most counties.

If you are unsure about identification and want to learn more, contact your local UF/IFAS Extension office and ask what parts of the plant to collect if you are asked to bring in a sample. If you are unable to collect a sample, then photographs of the plant can also be really helpful. Please be aware that even the experts can get stumped if you do not provide them with a proper sample or information on where the sample was collected.

Specialist Help for Solving Problems with Invasive Plants in Florida

In addition to contacting your local UF/IFAS Extension office, you have several options:

- You may submit a photo to the **UF/IFAS Distance Diagnostic and Identification System (DDIS)** online. URL: <http://ddis.ifas.ufl.edu>
- You may send questions to the **UF/IFAS Center for Aquatic and Invasive Plants (CAIP)**, Gainesville, FL 32653, Phone: 352-273-3667, E-mail: CAIP-website@ufl.edu
- You may call or write to the **Florida Fish and Wildlife Conservation Commission, Invasive Plant Management Section (Main Office)**, 3900 Commonwealth Blvd., MS 705 Tallahassee, FL 32399, Phone 850-617-9430, Fax 850-245-2835

First Point of Contact in States with Hydrilla Infestations as of 2014

ALABAMA (AL): Doug Carr, Alabama Department of Conservation and Natural Resources, Wildlife and Freshwater Fisheries Division, Aquatic Education Program, Phone: (334) 242-3884

ARIZONA (AZ): Arizona Game and Fish Department, 5000 W. Carefree Highway, Phoenix, AZ 85086-5000, Phone: (602) 942-3000

ARKANSAS (AR): Arkansas Game and Fish Commission, 2 Natural Resources Drive, Little Rock, AR 72205, Phone: (501) 223-6300, Phone (toll free): (800) 364-4263, Email: askAGFC@agfc.state.ar.us

CALIFORNIA (CA): California Department of Food and Agriculture, Plant Health and Pest Prevention Services, Sacramento, CA 95832, Phone: (916) 654-0768, Pest Hotline: (800) 491-1899, Email: ipcinfo@cdfa.ca.gov

CONNECTICUT (CT): Connecticut Agricultural Experiment Station Invasive Aquatic Plant Program (CAES IAPP), 123 Huntington Street, New Haven, CT 06511, Phone: (203) 974-8512

DELAWARE (DE): Delaware Invasive Species Council, Delaware Department of Agriculture, 2320 South Dupont Highway, Dover, DE 19901, Phone: (302) 698-4587, Email: disc@delawareinvasives.net

DISTRICT OF COLUMBIA (DC): Gina Ramos, Senior Weed Specialist, Bureau of Land Management, WO 220, 1849 Street, NW, Washington, DC 20240, Phone: (202) 912-7226, Email: gramos@blm.gov

FLORIDA (FL): University of Florida/IFAS, Center for Aquatic and Invasive Plants, 7922 NW 71 Street, Gainesville, Florida 32653, Information Office Phone: (352) 392-1799; or Florida Fish and Wildlife Conservation Commission, Invasive Plant Management Section (Main Office), 3900 Commonwealth Blvd., MS 705, Tallahassee, FL 32399, Phone: (850) 617-9430

GEORGIA (GA): Center for Invasive Species and Ecosystem Health, College of Agriculture and Environmental Sciences, University of Georgia, Tifton, GA 31794, Phone: (229) 386-3298

IDAHO (ID): Idaho Department of Water Resources, State Office, 322 East Front Street, Boise, ID 83720, Phone: (208) 287-4800, Email: IDWRinfo@idwr.idaho.gov

ILLINOIS (IL): Illinois' Hydrilla Task Force, Email: HydrillaHunt@niipp.net; or Illinois-Indiana Sea Grant College Program, University of Illinois, 1101 W. Peabody Drive, 350 National Soybean Research Center, MC-635 Urbana, IL 61801, Phone: (217) 333-6444, Email: iisg@illinois.edu; or Pat Charlebois, Aquatic Invasive Species Coordinator, Illinois Natural History Survey, Prairie Research Institute, c/o Chicago Botanic Garden, Phone: (847) 242-6441, Email: charlebo@illinois.edu

INDIANA (IN): Indiana Department of Natural Resources, Division of Fish and Wildlife, Indianapolis, IN 46204, Phone: (317) 234-3883, Email: dkeller@dnr.IN.gov; or Purdue University Extension, Purdue Plant & Pest Diagnostic Laboratory, West Lafayette, IN 47907, Phone: (765) 494-7071

LOUISIANA (LA): Louisiana Sea Grant College Program, 237 Sea Grant Building, Louisiana State University, Baton Rouge, LA 70803, Phone: (225) 578-6710

MAINE (ME): Maine Department of Environmental Protection (ME DEP), Phone: (800) 452-1942; or Maine Natural Area Program, Phone: (207) 287-8041

MARYLAND (MD): Maryland Department of Natural Resources, 580 Taylor Avenue, Annapolis, MD 21401, Phone: (877) 620-8367

MASSACHUSETTS (MA): Department of Conservation (DCR), Phone: (617) 626-1411 or (617) 626-1395

MICHIGAN (MI): Great Lakes Aquatic Nonindigenous Species Information System (GLANSIS), Toll-free phone: (877) STOP-ANS; or directly contact Rochelle Sturtevant, GLANSIS manager, Phone: (734) 741-2287, Email: rochelle.sturtevant@noaa.gov

MISSISSIPPI (MS): John D. Madsen, Mississippi State University, GeoResources Institute (GRI), Mississippi State, MS 39762, Phone: (662) 325-2428, Email: jmadsen@gri.msstate.edu

MISSOURI (MO): Missouri Department of Conservation Southwest Regional Office, Phone: (417) 895-6880

NEW JERSEY (NJ): Pat Rector, Environmental and Resource Management Agent, Rutgers, The State University of New Jersey, New Jersey Agricultural Experiment Station, Cooperative Extension of Morris County, Phone: (973) 285-8300 x225, Email: rector@njaes.rutgers.edu

NEW YORK STATE (NY): Roxanna Johnston, Watershed Coordinator, City of Ithaca, Phone (607) 273-4680, Email: roxannaj@cityofithaca.org; or Cornell Cooperative Extension Tompkins County, Ithaca, NY 14850, Phone: (607) 272-2292, Email: tompkins@cornell.edu

NORTH CAROLINA (NC): Aquatic Weed Control Program, Division of Water Resources, North Carolina Department of Environment, Health, and Natural Resources, Raleigh, NC, Phone (919) 733-4064; or North Carolina Department of Agriculture and Consumer Services, Weed Specialist, Phone: (800) 206-9333; or Eric Boyda, coordinator of the Appalachian Ohio Weed Control Partnership, Phone: (740) 534-6578, Email: appalachianohioweeds@gmail.com

PENNSYLVANIA (PA): Pennsylvania Fish and Boat Commission, Division of Environmental Services, Bellefonte, PA 16823, Phone: (814) 359-5147

SOUTH CAROLINA (SC): Aquatic Nuisance Species Program, South Carolina Department of Natural Resources (SCDNR), West Columbia, SC 29172, Phone: (803) 755-2872, Email: invasiveweeds@dnr.sc.gov

TENNESSEE (TN): Tennessee Valley Authority (TVA), Environmental Information Center, Phone: (800) 882-5263

TEXAS (TX): Texas A&M AgriLife Extension Service, College Station, TX 77843, Phone: (979) 845-7800, Email: help@agrilife.org

VIRGINIA (VA): Virginia Department of Game and Inland Fisheries, Richmond, VA 23230, Phone: (804) 367-1000, Email: dgifweb@dgif.virginia.gov

WASHINGTON STATE (WA): Washington Invasive Species Council, Olympia, WA 98501, Phone (877) 9-INFEST, Email: InvasiveSpecies@rco.wa.gov; or report online at www.InvasiveSpecies.wa.gov; or Jenifer Parsons, Washington State Department of Ecology, Yakima, WA 98902, Email: jenp461@ecy.wa.gov; detailed instructions at URL: <http://www.ecy.wa.gov/programs/wq/plants/plantid/mail.html>

WISCONSIN (WI): Wisconsin Department of Natural Resources (DNR), Madison, WI 53707, Phone: (608) 267-3531

Web Links to Hydrilla Fact Sheets

We have compiled a list of useful fact sheets that were developed by different state and federal agencies that you can find on the internet. Some are incorporated as pages into websites and others are in practical PDF format so you can print them.

Helpful Web Pages

UF/IFAS did not produce many of the publications listed here. The links are provided to help you find additional information. Pesticide application rules differ by state. Always refer to your state's Pesticide Information Office for application information.

BugwoodWiki. 2012. Hydrilla. Information developed by the Center for Invasive Species and Ecosystem Health at the University of Georgia. URL: <http://wiki.bugwood.org/Archive:BCIPEUS/Hydrilla> (23 July 2014).

BugwoodWiki. 2013. Hydrilla. Information developed by The Nature Conservancy. URL: http://wiki.bugwood.org/Hydrilla_verticillata (23 July 2014).

Invasive.org. 2003. Invasive plants of the Eastern United States. URL: <http://www.invasive.org/eastern/species/2626.html> (23 July 2014).

Invasive Species Specialist Group. 2006. Global invasive species database. URL: <http://www.issg.org/database/species/ecology.asp?si=272&fr=1&sts=sss> (23 July 2014).

National Park Service and U.S. Fish and Wildlife Service, Invasive Plants of the Eastern United States. 2003. Plant invaders of mid-Atlantic natural areas. URL: <http://www.invasive.org/eastern/midatlantic/hyve.html> (23 July 2014).

SEEPPC (Southeast Exotic Pest Plant Council). 2010. Hydrilla. Archive. URL: http://wiki.bugwood.org/Archive:SEEPPC/Hydrilla_-_Hydrilla_verticillata_%28L._f.%29_Royle (23 July 2014).

The New York Invasive Species Clearinghouse, Cornell Cooperative Extension Invasive Species Program. 2012. New York invasive species information. URL: http://www.nyis.info/index.php?action=invasive_detail&id=16 (23 July 2014).

USDA-APHIS (U.S. Department of Agriculture, Animal and Plant Health Inspection Service). No date. Federal Noxious Weed Dissemminules of the U.S. URL: <http://keys.lucidcentral.org/keys/FNW/FNW%20seeds/html/fact%20sheets/Hydrilla%20verticillata.htm> (23 July 2014).

Washington State Noxious Weed Control Board. 2010. Hydrilla. URL: <http://www.nwcb.wa.gov/detail.asp?weed=73> (23 July 2014).

Pesticide — a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals

Printable PDFs

UF/IFAS did not produce many of the publications listed here. The links are provided to help you find additional information. Pesticide application rules differ by state. Always refer to your state's Pesticide Information Office for application information.

FLEPPC (Florida Exotic Pest Plant Council) and University of Florida. No date. *Hydrilla verticillata* (L.f.) Royle. URL: http://www.fleppc.org/ID_BOOK/Hydrilla%20verticillata.pdf (23 July 2014).

FWC (Florida Fish and Wildlife Conservation Commission). No date. Division of Habitat and Species Conservation, Invasive Plant Management Section, Weed alert, hydrilla. URL: http://plants.ifas.ufl.edu/weedalert/invasiveplants_hydrilla.pdf (23 July 2014).

Hydrilla Task Force of the Cayuga Lake Watershed in NY State. 2013. Stop Hydrilla! URL: <http://ccetompkins.org/sites/all/files/347/stophydrilla.pdf> (23 July 2014).

Michigan Department of Environmental Quality. 2005. Heading off hydrilla. URL: http://www.nyis.info/user_uploads/files/Michigan%20DEQ%20hydrilla%20factsheet.pdf (23 July 2014).

Mississippi State University, Geosystems Research Institute. No date. Hydrilla. URL: <http://www.gri.msstate.edu/research/invspec/factsheets/2P/Hydrilla.pdf> (23 July 2014).

North Carolina State University, Agricultural Extension Service. 1992. Hydrilla. A rapidly spreading aquatic weed in North Carolina. URL: <http://www.weedscience.ncsu.edu/aquaticweeds/hydrilla.PDF> (23 July 2014).

State of Indiana. 2009. Aquatic invasive species. URL: <http://www.in.gov/dnr/files/hydrilla.pdf> (23 July 2014).

Washington Invasive Species Council. No date. Stop the invasion. URL: <http://www.invasivespecies.wa.gov/documents/priorities/HydrillaFactsheet.pdf> (23 July 2014).

Washington State Noxious Weed Control Board. 2013. Written findings of the Washington State Noxious Weed Control Board. URL: http://www.nwcb.wa.gov/siteFiles/Hydrilla_verticillata.pdf (23 July 2014).

Wisconsin Department of Natural Resources. 2009. Hydrilla. URL: <http://dnr.wi.gov/lakes/abcw/publications/WT-884.pdf> (23 July 2014).

FAQs and Stakeholder Feedback

Here are a few questions and concerns to consider when you are talking about and planning hydrilla management. We included several “real-life” comments we have received from various stakeholders, because we feel they deserve your consideration.

QUOTE: “At our HOA we added Asian grass carp at 7.5 per acre; the problem continued and we treated with Sonar; now we are considering additional Asian grass carp to bring it up to 10.5 [carp] per acre to control the new growth this year.” —J.M.

RESPONSE: J.M. correctly is expecting new growth in the year after successful herbicide treatment. It is important to remember that hydrilla forms tubers and turions that may remain viable (and undetected) in the sediment until the environmental conditions allow new growth. However, now that you have provided some control, the Asian carp that you already have in the water body may be able to keep up with the new growth.

Remember that Asian grass carp are very difficult to remove once they have been introduced. Too many Asian grass carp will result in removal of almost all plant material from the water body, which may also be undesirable depending on the situation. Contact the Florida Fish and Wildlife Conservation Commission for advice on stocking rates (see the Steps for Using Grass Carp section on page 40) specific to your water body and be aware that your permit for stocking grass carp is limited to the stocking rate that they recommend.

QUOTE: “I cannot evaluate submersed plants, but I have noticed a definite decline in the pickerelweed [after the release of grass carp]. I don’t know if there is a connection. I’m not aware that carp eat pickerelweed.” —R.S.

RESPONSE: The Asian grass carp is a generalist herbivore and readily feeds on aquatic plants other than hydrilla. Therefore, this biological control agent is best used in water bodies that need to be relatively free of aquatic vegetation, for example, in lakes with high boat traffic. In addition, the stocking rate (the number of Asian grass carp released per unit of water volume) is crucial to maintain a balanced relationship between the long-lived fish and the aquatic vegetation that is its food source.

QUOTE: “On our 250 acre lake, we have stocked triploid Asian grass carp in three installments and have mechanically harvested the weed when it was topping out—a couple of years ago. We have not used any other biologic control agents. There has been a dramatic reduction in the visible hydrilla, but this has been accompanied by a substantial reduction in clarity, presumably owing to algae benefitting from the increase in nutrients.” —R.S.

RESPONSE: When invasive plants, such as hydrilla, form monocultures in a water body and become topped out, the water clarity is often high. This is because the dense plant material is producing a lot of oxygen, blocking sunlight to the water column, and storing away nutrients so that they are not available for other organisms. Once the hydrilla is gone, the oxygen levels decrease, the sunlight can penetrate, and the nutrients are returned to the ecosystem. This allows algae to bloom. You have several options, but before you start, it might be a good idea to test the level of nitrogen and phosphorus to understand the problem better. The Green Industry Best Management Practices (GI-BMP) program has additional information you might find useful in your community. See page 29 for details.

Herbicide — a substance that kills weeds (usually a chemical compound)

Monoculture — the agricultural practice of growing one crop on a farm or in a production area

Submersed — a plant with most leaves growing underwater; flowers and some of the leaves may float on the water surface

Triploid [adj.], triploidy [n.] — when an organism has three sets of chromosomes; this is a rare condition in nature and leads to sterility in most organisms

Herbicide — a substance that kills weeds (usually a chemical compound)

Monoculture — the agricultural practice of growing one crop on a farm or in a production area



Important and useful information regarding plant management in Florida's waters is provided on the website of the UF/IFAS Center for Aquatic and Invasive Plants. URL : <http://plants.ifas.ufl.edu/manage> (Scan the QR code to connect to the website.)

First, examine the water body for potential sources of excessive nutrients. Reducing nutrient loading of the water body will improve lake health in the long-term. However, most water bodies in Florida naturally contain sufficient amounts of nutrient to facilitate plant growth and algal blooms without any external sources.

Second, it would be advisable to begin planting native aquatic plant species. Focus on plant species that the carp do not like to eat, such as spatterdock (*Nuphar advena*), eelgrass (*Vallisneria americana*), and fragrant waterlily (*Nymphaea odorata*), to prevent their consumption.

Third, manipulation of the nutrient availability is possible, particularly if the problem is due to excess phosphorus. Products can be added to the water to bind the phosphorus. See the Nutrient Management section on page 28. The type of algae that dominates will depend on the nitrogen to phosphorus ratio. If the level of nitrogen is low, then blue-green algae thrive; if the level of phosphorus is low, then green algae dominate. Green algae are better for the ecosystem than blue-green algae as they are a food source for many organisms. If blue-green algae and an excess of phosphorus are your problem and there are no options for controlling erosion from adjacent landscapes (the usual source of excess phosphorus), then manipulation of phosphorus level using the available products is a good option.

So you may be wondering if hydrilla control was the best option! Although a large biomass of hydrilla produces lots of oxygen, it also uses a lot of oxygen so on cloudy days the level of dissolved oxygen in the water may actually be very low, too low to support many species of plant, fish, or invertebrates. Topped-out hydrilla also prevents sunlight from reaching below the canopy preventing the growth of other submersed aquatic plants. A small area of hydrilla in your 250-acre lake would not be a problem, but hydrilla reproduces quickly and will soon spread. Once hydrilla has taken over, the number of species that can survive in the low-light, low-oxygen environment is limited. You will soon have a monoculture of hydrilla with little other flora.

QUOTE: “Last year I’ve seen herbicide sprayings that looked like they were getting paid by the gallon! Large dead masses in strange areas and dead cypress seedlings along river banks! Just think there must be a better way that benefits Florida’s wildlife. I see no benefit from massive sprayings other than opening to boat traffic? It’s just that after they spray, the area is void of life for quite some time.” —R.H.

RESPONSE: The decision to manage aquatic plants within a water body is never taken lightly. All herbicide treatments in public water bodies are regulated by the Florida Fish and Wildlife Conservation Commission (FWC). Any treatments in public water bodies or private water bodies that are linked to waters of special concern require a permit from the FWC. The FWC has specific guidelines that consider the impact on non-target species, with particular attention to endangered species. The FWC guidelines state:

“Application of herbicides shall be conducted at all times in a manner to cause the least possible adverse effect on human health, safety, recreational uses, non-target plants, fish, or wildlife.”

When choosing the most appropriate herbicide to use against hydrilla, the FWC biologists consider which herbicide will 1) provide the greatest protection to human health, 2) provide the greatest protection to non-target organisms, and 3) be most effective at controlling hydrilla. All aquatic herbicides are rigorously tested before they are granted registration by the U.S. Environmental Protection Agency and the Florida Department of Agriculture and Consumer Services. These tests include non-target toxicity testing to determine the products and rates that are safe to use in ecologically sensitive areas.

Systemic pesticides are often used for control of submersed plants as the effect of the chemical is not limited to the plant parts that contact the chemical. Instead, the chemical moves throughout the plant killing areas that were not exposed. For this reason, plants that grow near and take up herbicide-treated water are often effected and may sometimes be killed when this type of herbicide is applied. One example of an herbicide that will have this effect is fluridone: Trees and shrubs growing in water treated with this active ingredient may develop chlorosis or yellowing. However, chlorosis in non-target terrestrial plants is usually temporary, and the plants recover once the concentration of herbicide in the water declines.

It also is possible that R.H. described a foliar application made to emersed or floating vegetation growing in and around shoreline shrubs and trees as part of maintenance control. These treatments are often made using the contact herbicide diquat, which controls the targeted floating species and may defoliate non-target species like cypress. However, any cypress needles sprayed with diquat will regrow quickly, whereas the treated target species will remain absent. These maintenance control treatments may seem excessive, but they are highly targeted to small patches of invasive plants.

It is our responsibility as informed citizens to protect our environment. If you observe what you consider to be inappropriate use of aquatic herbicides, then you can contact the regional biologist at the Florida Fish and Wildlife Conservation Commission.

QUOTE: “I would like to see how [hydrilla management] affects birds and other wildlife and not a blanket statement that it does no harm. I would like to be constantly updated about how it affects the endangered snail kite. —J.W.

QUOTE: “I would like to see better and more consideration for the snail kite and other birds (eagles) who must forage during the winter months, especially since they seem to get going earlier these days. That is, do nothing during the early nesting season.” —S.W.

RESPONSE: J.W. and S.W. are not the only concerned citizens. It is important to be honest about potential side effects that a hydrilla management plan will have on non-target organisms, such as other aquatic plants and wildlife.

Any plan that is developed by the Florida Fish and Wildlife Conservation Commission for the management of hydrilla and other aquatic weeds in public waters includes considerations for non-target animal protection including snail kites. Although herbicides have no direct effect on snail kites or their prey, apple snails, there is concern over the effect of habitat loss and disturbance if hydrilla is controlled in snail kite nesting and foraging areas. Therefore, hydrilla control is not allowed during the period of snail kite nesting and peak juvenile foraging (a period that can extend from January to August).

An excerpt from the Everglade Snail Kite Brochure published by the Florida Fish and Wildlife Conservation Commission (FWC):

“Snail kites have been protected by state and federal law under the Endangered Species Act since 1967, making it illegal to harass, kill, capture or collect them. This includes protection from activities that disrupt normal breeding, feeding or sheltering.”

Habitat — an area or environment where a plant, fungus, or animal normally lives and grows

Pesticide — a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals

Submersed — a plant with most leaves growing underwater; flowers and some of the leaves may float on the water surface



Read the Everglade Snail Kite Brochure published by the Florida Fish and Wildlife Conservation Commission (FWC). URL: <http://myfwc.com/media/152978/Kites-Brochure.pdf>
(Scan the QR code to connect to the brochure online.)





Find an overview of the UF/IFAS GI-BMP Training and Program at the URL: <http://gibmp.ifas.ufl.edu> (Scan the QR code to connect to the website.)



Visit the following website to find the contact information of your local UF/IFAS Extension office. URL: <http://solutionsforyourlife.ufl.edu/map> (Scan the QR code to connect to a list of UF/IFAS Extension offices.)

QUOTE: “In the past, the FWC has taken the lead on the lake plants. [...] Our problem down here is the fertilizers! Both grazing and lawn chemicals, and sometimes both together - ‘improved’ grazing. [...] Since [the lake] is surrounded on the N, W, E by homes and grazing, I see no end to the problem.” —S.W.

RESPONSE: Florida is tackling this problem with the Green Industries Best Management Practices (GI-BMP) training program (see page 29). These guidelines were developed within the framework of Florida-Friendly Landscaping™.

Spread the Word

Everyone who visits water bodies should be aware of the problems caused by hydrilla and other invasive aquatic plants and of steps to prevent infestation and spread. Read on to get more ideas.

- Talk to your friends about invasive aquatic plant management.
- Bring up the topic at the next fishing tournament.
- Chat with people at the boat ramp when you launch into a fun day on the lake or river.
- Educate the public and have fun with it!

ARE YOU INVOLVED IN PUBLIC EDUCATION? Contact your local UF/IFAS Extension office for material you can use and for help with your programming. Each office has brochures and other educational print items available as well as PowerPoint presentations that you can use for events. Our team members will be happy to present our material or provide a display for aquatic weed workshops, field days, and other events to raise public awareness on invasive aquatic plants.

ALWAYS REMEMBER: Everyone who visits water bodies should be aware of the threats posed by hydrilla and other invasive aquatic plants and of the steps that are necessary to prevent infestation and spread.

ARE YOU PLANNING AN EVENT? We can loan you banners and provide you with educational materials that include 2-page brochures, 6-inch bookmarks (with ruler scale), web cards, and 14-page booklets (Figure 98).



Figure 98. Hydrilla IPM booth with banner displays, live plant and insect material, and educational items.



Find resources from the UF/IFAS Hydrilla IPM Project at the URL: <http://entomology.ifas.ufl.edu/hydrilla/resources.shtml> (Scan the QR code to see the resources online.)

Glossary

BIOTYPE: a form of the same plant species that shows special characters (for example, presence/absence of male or female flowers, resistance to a chemical herbicide, tolerance to extreme temperatures)

DIOECIOUS: female and male flowers occur on different plants

DIPLOID: a cell with two sets of chromosomes, usually one set is from the organism's mother and one set is from the organism's father; diploid cells are the building blocks for meiosis

DISTRIBUTION: the geographical range in which the plant occurs

HABITAT: an area or environment where a plant, fungus, or animal normally lives and grows

HERBICIDE: a substance that kills weeds (usually a chemical compound)

HERBIVOROUS [ADJ.], HERBIVORY [N.]: plant-eating

INDIGENOUS: occurring naturally in a place or region

MEIOSIS: when a diploid cell divides into haploid cells to produce reproductive gametes (this is the process required to produce sperm and eggs or pollen and ovules)

MONOCULTURE: the agricultural practice of growing one crop on a farm or in a production area

MONOECIOUS: female and male flowers occur on the same plant

NUCLEUS (CELL NUCLEUS) [SING.], NUCLEI [PL.]: a membrane-bound organelle found in most cells; this organelle contains most of a cell's genetic material, and it is where the chromosomes are located

PATHOGENIC: disease-causing

PESTICIDE: a substance that is used to destroy insects or other organisms that are considered harmful to cultivated and native plants or animals

pH: a term used in chemistry to indicate the acidity or alkalinity of a solution based on the availability of free hydrogen; values range from 1 to 14; pH 7 = neutral, low pH (1 to <7) = acidic, high pH (>7 to 14) = alkaline

RESISTANCE [N.], RESISTANT [ADJ.] (TO HERBICIDES): the ability of a plant to survive the exposure to a typically lethal dose of herbicide

SUBMERSED: a plant with most leaves growing underwater; flowers and some of the leaves may float on the water surface

SYNERGISTIC EFFECT: the effect caused by two methods or agents together is greater than the sum of the effects of each individual method or agent

TERRESTRIAL: relating to the earth, for example, an organism that lives on land (as opposed to one that lives in water)

TRIPLOID [ADJ.], TRIPLOIDY [N.]: when an organism has three sets of chromosomes; this is a rare condition in nature and leads to sterility in most organisms

WHORL (LEAF WHORL): an arrangement of three or more leaves emerging and radiating from a common node along the stem

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