Aquatics

JUNE 1986

EDITORIAL

"Out of sight — out of mind" and "The Squeaky Wheel Gets the grease" are familiar little expressions with big implications in today's aquatic plant management in Florida.

It has been but a few years since Florida's St. John's River, Orange Lake and other prestigious waters were heavily dominated by exotic aquatic weeds. These weeds impeded navigation and all but stopped fishing, yet effectively united the effort toward managing our state waters. Concerned water users gathered political support and a trust fund was established under DNR to be used with federal funds to manage and control aquatic problems. Gradually, as a result of hard work and constant vigilance, the overwhelming problems have been brought under control.

The 'good ole days' in aquatic plant control were only good for the plants. Fish, navigation, crops and everything else suffered. So what's the big deal? These waters are being properly managed and the funds will continue to support the maintenance control activities. Right? Wrong! Read On!

Last year an exceptionally well-greased bill slid through the legislature which in April, 1986, removed 1.5 million dollars from DNR's aquatic plant control funds. A project identified as one to clean up Lake Apopka by utilizing water hyacinths for muck removal is the legislative leach. Interested state politicians supported the legislation to create the Lake Apopka Restoration Council which directs this project. Although funded by DNR and involving aquatic plant research — neither DNR nor the aquatic weed research community are represented on the council.

Last year after concern was voiced to the Governor's Office, letters to FAPMS assured all that future funds would not *come from DNR. This money is imperative if weed control operations are to continue in Florida's lakes, rivers and canals. DNR's Aquatic Plant Trust Fund was not established to fund lake projects involving muck removal using exotic weeds. DER is charged with funding such*

continued on inside back cover

ABOUT THE COVER

A lazy summer afternoon on McKeathan Lake
Photo by: Jim Kelley

Aquatics

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AQUATICS: Published quarterly as the official publication of the Florida Aquatic Plant Management Society. This publication is intended to keep all interests informaed on matters as they relate to aquatic plant management particularly in Florida.

EDITORIAL: Address all correspondence regarding editorial matter to Daniel Thayer, Editor, "Aquatics" Magazine, 7922 N.W. 71st Street, Gainesville, FL 32606.

The Fringed Bog Orchids

by

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"To spray or not to spray, that is the question. Whether 'tis nobler to suffer the slings and arrows of outrageous weeds or to spray back and by so spraying end them." — Shakeysprayer

Introduction and Taxonomy

With all due apologies to Shakespeare, to spray or not *is* often the question. When airspeed, season, water temperature or other factors are unfavorable, proposed weed control has to be aborted or amended. Special concern must be given, also, to impact of any control operation upon the locality since non-target damage should be avoided. Such damage can induce new growth of some hardy species, with regrowth periods being fairly brief, yet overspray of any of the aquatic orchids of the Habeneria genus can eradicate these sensitive species. Furthermore, such eradication could constitute violation of federal law, namely the Endangered Species Act. Five native members of the genus are federally listed as threatened species. They are *H. distans, nivea, odontopetala, quinquesta* and *repens.* Seventeen members of the genus are recognized in the Southeastern United States.[1]

The Act defines "threatened species" as those "likely to become . . . endangered species within the foreseeable future throughout all or a significant part of their range."[2] Violations of the provisions of the Endangered Species Act are civil infractions subject to maximum $10,000 fines. All five listed species occur in Florida. However they never constitute more than a minor component of a given plant community. Typically called the fringed, bog or creeping spider orchid the semiaquatic species most commonly encountered in Florida is *H. repens* Nutt.

Fringed Bog Orchid (Habemeria repens)

Habitat and Structure

Orchidaceae world-wide number over 30,000 species and are perennial with terrestrial, epiphytic or semiaquatic growth habits. The Habeneria would all be termed semiaquatic or terrestrial. The semiaquatic species are often encountered as loosely rooted individual or colonial groups growing amongst shoreline and floating tussock-forming plants. In Florida these are often sedges *'Cyperus* spp.), water-hyacinth *(Eichhornia crassipes* [Mart.] Solms.), smartweeds *(Polygonum* spp.) and evening primroses *(Ludwigia* spp.). Typically, this community is found in the litteral zone, yet can establish free-floating mats. The bog orchids insinuate themselves within this plant community and gain support from the dense root mat and other plants' emergent foliage. The surrounding vegetation also absorbs wave action, shielding the lightly rooted orchids.

Habeneria species have an erect growth habit with simple, fleshy roots, basal leaves sheathing the stem and many small flowers borne on a single terminal stalk or raceme. Key characters in the differentiation of the several species include the elaboration of the flower petals. The genus name stems from the Latin *habena* (rein), referring to the sour-like structure at the base of the flower lip. Florida's members of the genus have pale green to cream-colored flowers. The plants reach about two feet in height.

Reproduction and Distribution

All orchids are monocots whose

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Continued from page 4

flowers are sexually complete with the male and female structures fused into a column. Also, one petal is usually exaggerated into a bulging pouch-shaped lip that often supports a nectary. Sexual reproduction is effected by insects (often moths among the Haberneria) transporting pollen from the anther to the pistil. Flowering on a stalk begins nearest the base and proceeds to the unopened buds at the tip. In this way flowering on any stalk can continue for many days. Numerous tiny seeds develop within the fertilized ovule. Transportation to new sites can occur by wind, water or animal actions. Once established a mature plant will asexually produce identical offset plants so that a substantial clump or colony of genetically identical plants can arise. These plantlets arise from the parent's roots.

Bog orchid species occur from Ontario into Florida panhandle lakes and on to Everglades hammocks and Caribbean tropics. Usually the soils and waters in which Haberneria species will flourish tend to be fairly acidic and soft. Bodies of water which support them generally would be classed as oligotrophic to mesotrophic. It is in just such waters that diversity of species would be expected to be highest. In such waters competition for a few available nutrients may be great but usually more species participate in the game.

The Department of Natural Resources aquatic plant survey publications have reported *Haberneria repens* in only three Florida lakes with a maximum coverage of one acre statewide. I've become aware of the plant in seven lakes in one county alone, yet this was in 1985 when the DNR survey was published with data on solely the BIG THREE: water-hyacinth, water-lettuce, and hydrilla. Doubtless *H. repens* occurs in many more lakes, yet due to an inconspicuous growth habit it has escaped inclusion. All available information indicates that the Habeneria species do not survive in rivers.

Conclusion

Inconspicuous species of rare occurrence do not normally generate much concern or notice until they're gone or nearly so. (Much of the same could be said of that lemon on your left until you notice that he's disappeared with your wallet.) Haberneria species are not showy orchids like those your aunt has all over her porch. They do not support a vast community of critters that will live on no other plant nor do they absorb lots of nutrients or serve humanity in a well-defined way. They may only be missed when spray drift lands on the last individual. And that will be too late.

1 Godfrey, Robert K. and J.W. Wooten. Aquatic and Wetland Plants of Southeastern United States, University of Georgia Press, 1981, 627-47.

2 The Endangered Species Act, Public Law 94-325. 1973.

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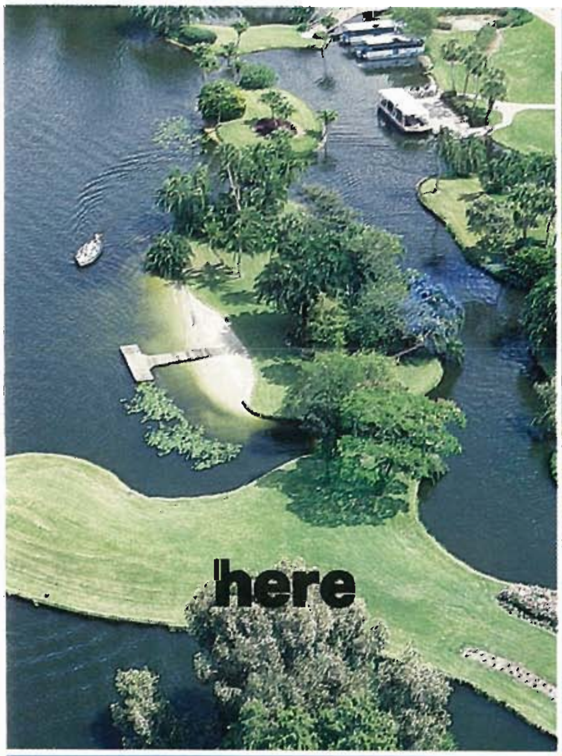

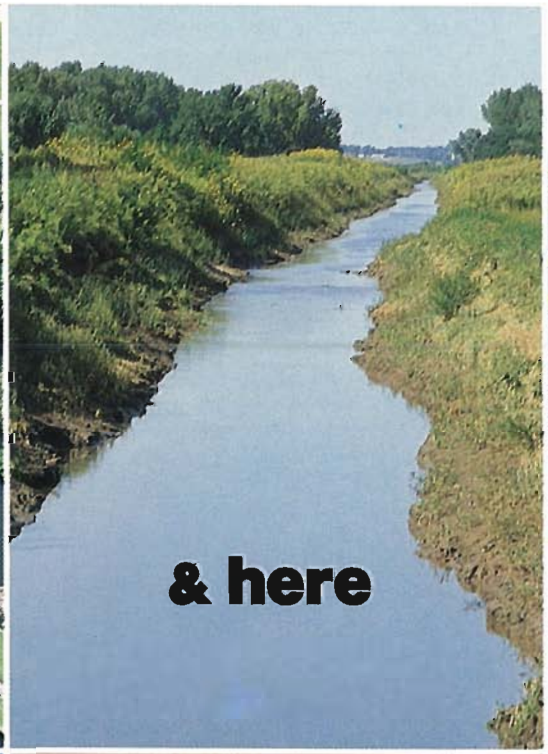

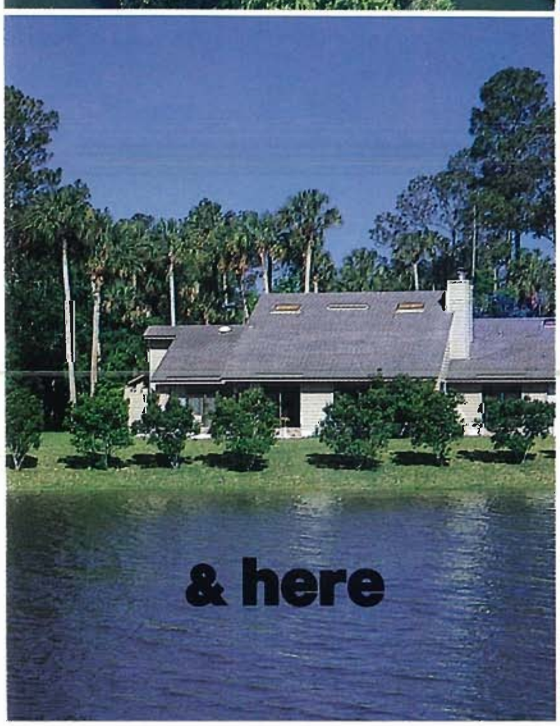

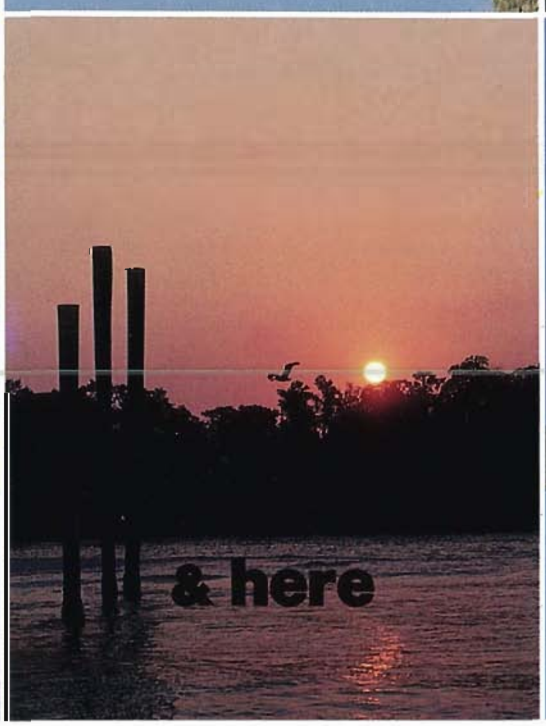

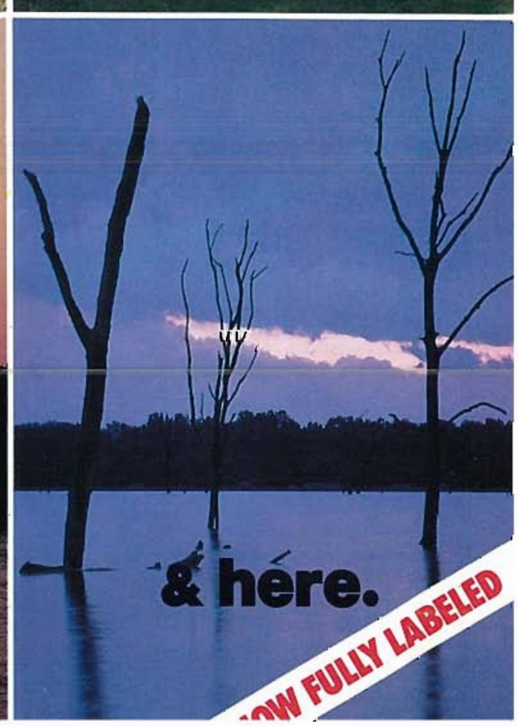

DEATH IN THE DOSE

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Toxicologists have long known it, and educated laymen give it lip service: The Dose Determines the Poison. In other words, there's nothing so toxic that in small enough amounts would not be completely harmless to a living organism; and there's nothing so safe that in large enough amounts would not be fatal to that creature.

The ramifications of this scientific truism should be important to the entire agrichemical industry. It should be a moderating influence on the trend to regulate pesticides to the ever more minute particle. It should be ample evidence to decrease liability both morally and legally. It should give good cause to emend or strike the Delaney clause, which prohibits any residue, no matter how tiny, that has been shown to be carcinogenic. It should encourage us to doubt the applicability of animal-cancer tests that rely on wild extrapolations from mouse to man.

But, in fact, it does none of these things. The populace and the lawmakers remain oddly convinced or at least unmoved by the maxim that dose determines poison.

It seems right to ask why science carries no weight in this case. The easy answers would be that the agrichemical industry gets distorted media coverage or that there is prejudice against it. But these answers really beg the question. Why is there this prejudice in the first place? The real answer must go deep.

And deep means into the psyche. Neil Orloff has proposed three psychological reasons why Americans fear pesticides far out of proportion to the real, scientifically determined risk. Orloff, who is director of Cornell University's think tank Center for Environmental Research, postulates these underlying causes at play in the American psyche. First, there's the emotional need to find a scapegoat for dreaded diseases like cancer. Second, there's the ingrained belief that nature and its products must be essentially pure and wholesome. Third, there's a pent-up need to lash out at big business to release our frustrations and resentment.

If you ponder these three motivations, you'll realize why hard, cold facts just don't cut the mustard when it comes to pesticides. The cold reality, for example, of the "death is in the dose" maxim undercuts the belief that nature is pristine and always benign while man-induced pollutants are the only corruptors. The fact is that nature as such is fundamentally neutral in this regard. It supports our life, yes, but its elements are equally effective as poisons to us. Hence the toxologists' saying runs counter to the heart-felt prejudices and feelings that most of us share. It's no wonder, then, that merely strutting scientific facts does not seem to be winning either opinion polls or court room cases nowadays.

Let's take a recent example. Orloff noted that officials have been damned when they try to placate crowds by stressing that a risk is only "one in a million." A recent in-house newsletter by Monsanto similarly tried to quell employee's concerns over the special review of Lasso by noting that the potential risk from a lifetime exposure to Lasso is about "one in one million." To make its point, the article explained in graphic terms just what one in a million means by comparing the risk to that of smoking 1.4 cigarettes once in a lifetime, drinking 30 cans of soda containing saccharin, receiving one properly administered X-ray, or being exposed to the added radiation form two months living at a high altitude.

All of Monsanto's examples make the point well that we gladly take one-in-a-million risks everyday. But will this convince everyone that they should therefore take that same risk with a pesticide?

My Trip to Pakistan and India — Weeds, Weevils and Worries

by
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Here we are, two Americans sitting in a dried up pond on a hot Indian evening picking insects out of our mouths, eyes and nose as we stare at a black light waiting for the right insect to land. People from nearby houses help us pick up insects from the sheet. They laugh and tell jokes about crazy Americans. Why, you ask, would anyone travel thousands of miles to sit in a dried up pond? The reason any dedicated entomologist does most things is to collect insects. Specifically we are looking for two hydrilla feeding insects. If successful, we would take the insects home to Florida and eventually use them for biological control of hydrilla.

In April 1985, Gary Buckingham, Research Entomologist, ARS-USDA and I traveled to Pakistan and India to search for and collect the fly, *Hydrellia pakistanae* and the weevil, *Bagous affinis*. The main purpose of our trip was to collect the fly, but we also had some questions from our laboratory studies about the weevil that needed field confirmation. The trip was preceded by months of planning and preparation. We wanted to reach Bangalore in southern India at the end of the dry season, just before the monsoons started because there would be less water and the insects would be concentrated. Official visas and travel authorizations had been requested three months before departure. Health department requirements had to be learned, five shots coordinated, and malaria prophylaxis obtained from a private physician. Contacts in both countries had to be notified and local arrangements made. Ten boxes of equipment had to be assembled, packed and sent.

Our principal target, *Hydrellia pakistanae* is a small leaf mining fly, about one-fourth the size of a mosquito, in the family Ephydridae. The biology is summarized here from research done in Pakistan by Baloch and Sana-Ullah (1973) and Baloch et al. (1980). Flies mated in the laboratory shortly after emergence and oviposited a day later. The eggs were laid singly or in small groups on floating vegetation. The larvae hatched in three to four days and entered and started mining a leaf. After mining all the leaves in one whorl, the larvae then moved to another whorl. Each larvae can damage an average of 12 (10-17) leaves. The larval stage lasted nine to 16 days and the pupal stage six to 11 days. In the field there were about six generations per year.

Mr. Krishnaswamy, left, and Mr. Narayan Nair collecting hydrilla infested with the fly.

Our other target, *Bagous affinis*, attacks the subterranean turions (tubers) of hydrilla. The adults feed on hydrilla stems and turions as the water dries up. In the laboratory, the females preferred to lay their eggs in moist, soft wood, but in the field the eggs are probably also laid in the soil. Females laid an average of 232 eggs. Larvae hatched in three to four days and then crawled through the soil to burrow in turions. The larvae which are cream color with brown heads, fed inside the turion scooping out the insides. Generally, there was only one larva per turion, but two or more larvae per turion have been observed both in the lab and the field. The larval stage lasted eight to 10 days and the pupal stage four to six days at 27 C. Pupation took place inside the turion. After emerging, soft adults sit in the turion or in moist soil for one or two days to finish hardening. Adults live up to nine months in the lab, although most died within a couple of months. The larvae in turions died when totally submerged for 96 hours, so *Bagous affinis* would not be an effective control agent except when the turions are exposed.

Our first stop was Rawalpindi, Pakistan. We arrived safely but our luggage including our collecting equipment was missing for the next five days. Rawalpindi is an old city with small crowded streets. Next door to it is the newer, more modern capital, Islamabad. While in Rawalpindi, we visited the Commonwealth Institute of Biological Control (CIBC) laboratory. CIBC is an international organization which conducted projects on hydrilla and water milfoil in the 1970s for the United States Department of Agriculture. Most of the scientists and their assistants who worked on the initial project have since left the laboratory. However, one of their drivers/field collectors remembered the collection sites and was able to take us to the exact spots where the two insects had been previously collected. With his help we were immediately successful, whereas without it we would have spent many days searching. We collected at three locations in the Rawalpindi area and evidence of the fly was found at all three locations. The largest populations were in several small drying ponds in Ayub National Park. Most stems had a few damaged leaves but there was no extensive damage. We were pleasantly surprised to find mines of the fly on hydrilla about a foot underneath the water's surface in a deep waterhole in a dry river bed. We had previously thought that the fly attacked only topped-out hydrilla. Apparently the larvae had dropped to the plants from eggs laid at the water's surface. We found only one attacked hydrilla turion and no weevil adults, although we did not observe many turions, which were less numerous than in our Florida lakes. Pondweed turions were also checked for weevil damage, but none were attacked. Before departing Rawalpindi, we placed the hydrilla containing *Hydrellia* larvae and pupae in jars at the CIBC lab. We hoped this material could be shipped to Gainesville, if no insects were found later in India.

We were fortunate to be able to ride to Lahore with two CIBC workers where they had integrated pest management research plots in orchards. Lahore, in the famous Punjab Province, is a fairly modern city that was once a favorite vacation site of the British. Although Lahore is only 150 miles from Rawalpindi, the trip took nine hours to drive. The trip was a real adventure. The road, by our standards, was narrow and rough. The driver was always dodging ox carts and pedestrians, not to mention other cars, trucks and buses. Little did I know the worst was yet to come. Indian roads made Pakistani roads look deserted. At a drying pond along the way near Nadipur, we finally found a large number of attacked turions. The five of us spent about an hour in the hot sun searching the roots of cattails, sedges, pondweeds, and various unidentified plants for weevil damage. None of the roots were attacked. Another former CIBC collecting site about 10 miles N. of Lahore also yielded infested hydrilla turions. There was little water remaining at either place, and no evidence of the fly was found.

From Lahore, we flew to New Delhi, India. We arrived in New Dehli, to very hot and dry weather, but at least our luggage was with us and someone met us at the airport. We spent the next five days meeting with officials of FERRO, the Far Eastern Regional Research Office, OICD, UDSA at the American Embassy. With their help, we received our travel advance and official sanction of our trip by the Indian Government. We also visited Indian scientists who discussed their research with us.

We left New Delhi and flew to Bangalore in the south Indian state of Karnataka. Bangalore is a modern city and was less congested than New Delhi. The climate is also cooler and was a welcome change. We chose to go to Bangalore because of the CIBC laboratory located there. CIBC personnel had previously sent us shipments of the weevil, and one of their scientists, Mr. Krishnaswamy, had started a small colony of the fly right before we arrived. They provided us with a small greenhouse where we could set up our equipment and work for the next twenty days. Mr. Krishnaswamy, and their driver/field collector, Mr. Narayan Nair, took us to their collection sites and helped us with our field work. Without their help and our other contacts both in India and in Pakistan, our job would have been more difficult and not nearly as successful.

We collected at three different locations near Bangalor. The first collection site consisted of three or four small partially dried ponds on the road to the historic city of Mysore. The other two sites were large, shallow lakes. One of the lakes, or "tanks" as they are called in India, covered 6 acres, but by the end of the dry season contained less than an acre of water. The fly was common at both lakes, and at other areas where there was sufficient water and hydrilla. However, the plants were not heavily damaged. Most of the damage we noticed was to the leaves at the tips of the stems. One reason why the fly was not as effective could have been natural enemies. We found at least three different parasitic wasps that emerged from the leaves. About half of the fly larvae I brought home were parasitized. Another problem was that any lake, including our two, with enough water and plants for the fly would be used by local villagers for fishing and irrigation. The first thing they would do is pull the plants out and throw them up on shore. This practice may also help reduce the fly populations.

Besides collecting during the day, we spent three evenings running a black light similar to the lights used in mosquito zappers. The first two nights, we worked at a pond that had dried up except for a few deep holes. Because batteries are scarce in India we had to use the one in our Jeep. This meant driving the Jeep out on the dry pond and avoiding the pot holes. There were a few times as we were leaving in the dark, when I thought we were going to get stuck in the holes. We had predicated from our laboratory experience that the weevils should be at these dry ponds now, and would fly to a blacklight, but our CIBC companions were skeptical. We were so excited that first night when the first weevil landed on the sheet right after dusk, and proved us right. The four of us, along with the help of nearby villagers collected many of our weevils and other insects. The second night was a disappointment. Right as the insects should have started flying, a storm ended our blacklighting. The last time we collected at a different location. Our weevils along with many other aquatic insects just poured into the light by the thousands, covering us, the collecting sheet and the ground.

We usually did our field work in the morning to avoid the hot afternoon sun, and then returned to the laboratory for a lunch of mangoes and cookies; no fast food restaurants in India. Afterwards we processed our many plastic bags of material. The greenhouse was a nice place to work. We never knew when we were going to be entertained by an unusual Indian bird or the troop of monkeys that lived on the grounds. Mr. Krishnaswamy took care of the fly while Gary and I checked hydrilla turions for weevil larvae and pupae using the microscope we had sent over. We found many attacked turions along with some larvae and some pupae. The eggs from the weevils collected with the blacklight were used in an experiment with Indian hydrilla turions using our Florida methods and test materials which we had sent over. The results of these tests were to be compared to results of experiments we had already done at home using Florida turions and the same methods and materials. The Indian turions were sparse and difficult to collect because of the rock-hard soil. The most we ever collected at one time was 102, and that was with four of us digging. All our hard work was almost for nothing since our collections were nearly wiped our one night by a hungry rat which chewed into our rearing cups to get the infested turions.

Before we left Bangalore for home, the insects had to be packed for the journey. Past shipments of the weevil by the CIBC lab had been successful, but we had no experience shipping the fly. Heavy plastic bags with screw tops that we had carried with us to India were filled with water, hydrilla sprigs and fly larvae and pupae. We tested these in darkness for one day at the CIBC before making the final decision. One day was to be the longest period that would be in the luggage. The bags were carried in my hand luggage and removed at each hotel so the plants could get light and replenish the oxygen supply. The bags worked well for the flies but not so well for my suitcase, since they leaked. The trip took three days and at the last hotel in Frankfort, Germany, the plastic bags were sealed in heavy cloth bags for the final leg home. The weevils were carried in plastic containers with moist blotter paper, wood excelsior and hydrilla. The containers were placed in cloth bags at the CIBC.

Both species of insects, and I, successfully survived the trip. The weevil adults were added to our established quarantine colony to increase the genetic variability and the fly was successfully colonized in quarantine. We have initiated host range tests with the fly. Arrangements were made before we left Bangalore for Mr. Krishnaswamy to conduct field surveys of native plants for *Hydrellia*, and to send more insects if they are needed. Our trip answered several of our questions about *Bagous affinis* and we now plan to petition for permission to release it.

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The author and helpers collecting tubers in India.

Algal Identification

by
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The term ALGAE describes a diverse group of plants that range from microscopic unicellular forms to 300 foot seaweeds. Although some algae may appear fairly complex in structure, they lack the elaborate tissue anatomy and reproductive structures of the more "advanced" plants. Technically defined, algae are thallophytes (plants lacking roots, stems, and leaves), have chlorophyll *a* as their primary photosynthetic pigment, and lack a sterile covering of cells around the reproductive structures. Thus, algal zygotes (spores) are usually released directly into the water once fertilization is complete. Since algae are nonvascular (lack water and food conducting tissues) all cells of the organism must be exposed to water and nutrients in order to live. This also means that an algicide applied to water must come in contact with all the cells to be effective, since there is no conducting tissue to move the chemical to other parts of the organism. This is one reason why good covereage of algal mats with copper is important to good treatment.

There are at least 9 major groups or divisions of algae. Among these are the: Cyanophyta (Blue-greens), Chlorophyta (Greens), Euglenophyta (Euglenoids), Chrysophyta (Golden browns and diatoms), Pyrrophyta (Dinoflagellates), Phaeophyta (Browns), and Rhodophyta (Reds). As you can see, the basis for these names is primarily color or pigmentation. However, using color to identify algae doesn't always work in the field because (for example) some blue-green algae are red, some red algae are green, and some golden browns are yellow green!

Nonweedy Algal Groups

The brown and red algae are the seaweeds of coastal areas. The browns, commonly known as the kelps, are strictly marine; there are no known freshwater species. The reds are primarily found as seaweeds but a few species are freshwater (for example, *Bangia* found along the shoreline of Lake Michigan). However, they are seldom if ever found in concentrations dense enough to require treatment.

Although frequently found in the freshwater plankton, dinoflagellates also are seldom problems. Freshwater dinoflagellates are unicellular and golden brown in color. They sometimes bloom in sufficient quantities to produce a brown stain to the water but are more generally found intermixed with other kinds of algae. Marine species of dinoflagellates form the infamous "red tides". Their blooms stain the coastal waters red and a toxin produced may kill fish and other organisms. When taken in by shellfish, the toxin is concentrated and is lethal to any warmblooded organism (including humans) that consume the shellfish. The shellfish itself is not affected. Once the bloom subsides, the shellfish are cleansed of the toxin and are safely eaten again. The shellfish industry of the east coast is disrupted almost every year (usually in the fall) because of red tides.

A common dinoflagellate in the plankton is *Ceratium* (Figure 2). This alga is easy to identify with a microscope because it is so large and unusual in shape. It is golden brown in color and swims.

Major Weedy Algal Groups

The other groups of algae are much more important as aquatic weeds. The three major growth forms are microscopic, mat-forming, and charoid. The matformers and charoids are, like flowering plants, part of the macrophyte flora.

Blue-green Algae

The blue-greens are usually considered to be the most noxious of algal growths because of their ability to produce dense blooms in nutrient-enriched waters. Some strains are toxic to fish and warmblooded animals, but their major harmful effect is in causing oxygen depletion when the blooms crash. Over a period of years, blooms and crashes of blue-greens can lead to the development of anoxic conditions in the bottom waters of a lake.

The blue-green species that cause blooms are all microscopic. Other blue-greens are mat-formers that usually begin growth on the bottom sediments but then float to the surface as black, slimy blobs. One species, *Oscillatoria rubescens*, is red in color and is found under the ice in winter. It may turn a lake red at the spring turnover but then subsides into the deeper, cooler waters as the season progresses.

There are two important features to remember about the blue-greens in comparison to members of the other algal groups. First, they have extremely small cells. The cells are usually aggregated into colonies or filaments; single cells are seldom found floating freely in the water. In addition, the colonies or filaments are often encased in a thick slime sheath. The large size of these colonies plus the slime enables blue-greens to avoid predation by zooplankton. The second feature of blue-greens is the presence of pigments in addition to chlorophyll *a*. All have phycocyanin (blue) and phycoerythrin (red) pigments. In addition, the sheaths may pick up metallic compounds or debris from the water that give color. The main point is that blue-greens seldom appear blue-green. Colors may range from black to purple to light green, depending on the intensity and combination of pigments.

Microscopic blue-greens. Blooms of the "big three", *Anabaena, Aphanizomenon,* and *Microcystis* (Figure 1) usually give the water a pea-green to yellow-green color. Under calm conditions, they form scums at the water surface. This is due to the presence of internal gas vacuoles that make the cells extremely bouyant. When these organisms are observed under a microscope, they no longer appear green but will look dark brown or even black. In this case, the color is not due to pigment but to the light that bounces off the gas vacuoles. The greater the abundance of gas vacuoles the darker brown the color of cells is under the microscope.

Blue-greens are the only algae with gas vacuoles. However, not all blue-greens have gas vacuoles. Some planktonics do not, nor do the majority of the mat-formers.

Mat-forming blue-greens. These are primarily in two genera: *Oscillatoria* and *Lyngbya* (Ling-bee-ah). The mats are usually very slimy and appear a dark blue-green to black or brown. They seldom form a surface mat of vegetation. They more typically grow on the bottom and float to the surface temporarily when dislodged by wind, wave, or animal activity. *Lyngbya* is the most frequent cause of "black algae" growths in swimming pools. The two genera are similar in appearance under the

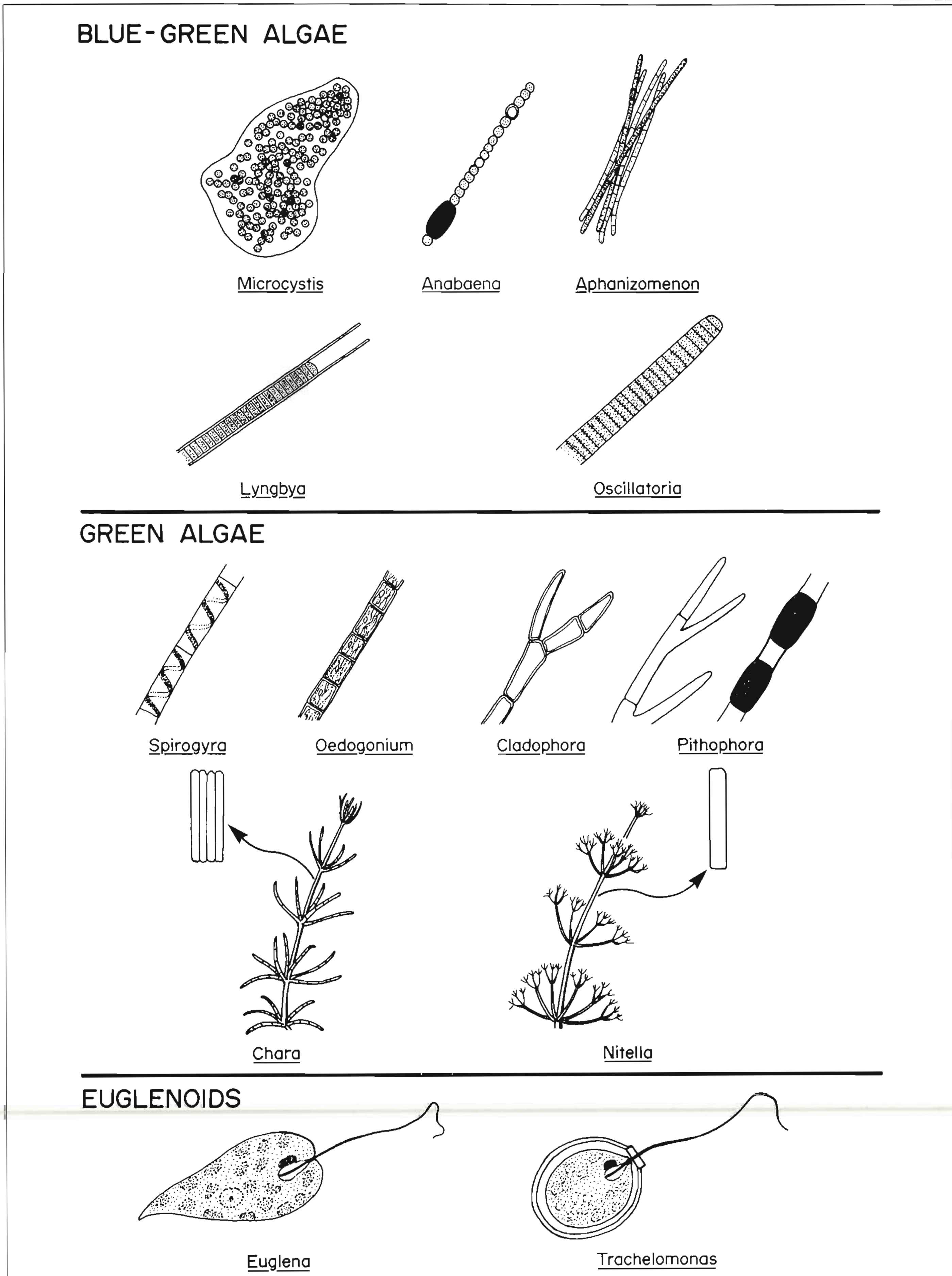

Figure 1. Algae that are primarily green in color. Blue-greens often appear black, brown, or red and the euglenoids can cause red or red-brown blooms.

microscope except that *Lyngbya* has long cell wall structures that extend beyond the filament (Figure 1). True to its name, the filaments of *Oscillatoria* usually oscillate (sway back and forth) when viewed under a microscope.

Green Algae

Unlike the blue-greens, the greens are indeed green in color. All three growth forms are found in the green algae. Many are microscopic, consisting of single cells or small colonies. Although they may cause planktonic blooms, they are considered less troublesome than the blue-greens. Some common genera of microscopic greens are *Scenedesmus*, *Chlorella*, and *Pediastrum*. The mat-forming and charoid greens are the most common of the weedy algae.

Mat-forming greens. These generally form conspicuous attached or free-floating mats of algae. The color is green turning yellow-green as the mats get older. The four major genera are *Spirogyra*, *Oedogonium*, *Cladophora*, and *Pithophora* (Figure 1). In the field they can be distinguished by texture. *Spirogyra* is extremely slimy, *Cladophora* is cottony, and *Pithophora* is stiff and coarse. *Oedogonium* (ee-doe-gon-ee-um) is somewhere between *Spirogyra* and *Cladophora*. The major features used to identify these algae are listed in Table 1.

Table 1. Characteristics of four common filamentous green algae.

	Spirogyra	Oedogonium	Cladophora	Pithophora
Habitat	Free-floating	Free-floating; often clinging to macrophytes	Attached or free-floating	Free-floating
Seasonality (Midwest)	Most abundant in spring and early summer	Probably most of the year	May and June persisting to mid-summer	July through October
Texture	Slimy	Intermediate	Cottony	Coarse
Visible features	Bright green	Yellowish green	Branched	Branched with akinetes
Microscope	Unbranched	Unbranched	Branched	Branched
	Long cells	Short cells	Long cells	Long cells
	Spiral chloroplast	Overall green color	V-branching	Branches laterally

**TABLE 2.
Characteristics of the charoid algae.**

Chara	*Nitella*
Usually calcified	Not usually calcified
Leaves unbranched	Leaves branched, often many times
"Stem" is corticated; internodal cell surrounded by layer of cells. (See Fig. 1)	"Stem" noncorticated; internode consists of a single cell. (See Fig. 1)

Charoid greens. Charoids are the most "complex" of the freshwater algae and resemble flowering plants in their overall appearance. They have minute rhizoids that anchor them into the sediments, and stem and leaf-like appendage. However, there is no conducting tissue and each cell must be exposed to algicide in order to be effectively killed. The two genera are *Chara* and *Nitella* (Figure 1). *Chara* is a more frequent problem than *Nitella* because it can tolerate alkalinity and high pH that is typical of many of our waters. The majority of *Nitella* species are found in waters of pH 5 to 7 and with calcium concentrations of less than 100 mg/1. The apparent reason for this is that *Nitella* requires CO_2 as a carbon source for photosynthesis. As pH and water hardness and alkalinity increase, CO_2 concentrations decrease and bicarbonate becomes the most abundant form of carbon. *Chara* found in waters of pH 7 to 9 and calcium concentrations of 100 mg/1 apparently can switch to bicarbonate when CO_2 becomes limiting; *Nitella* can not. This does not mean that *Nitella* is never found in alkaline waters. It can be, for example, attached to seawalls where wave action insures its exposure to atmospheric CO_2. It certainly is a problem in localized areas with acidic to neutral waters although it tends to form shorter plants that *Chara*. Table 2 lists the characteristics of charoids.

Euglenoid Algae

All of these algae are unicellular and microscopic. They have a single flagellum and swim. Although most are green, their major feature as aquatic weeds is that some species are a common cause of red water. They are particularly common in organically polluted water (e.g., in waters receiving feed lot runoff) since they can switch from photosynthesis during the day to the utilization of organic compounds at night to derive energy (this is one reason why euglenoids are often considered animals by zoologists).

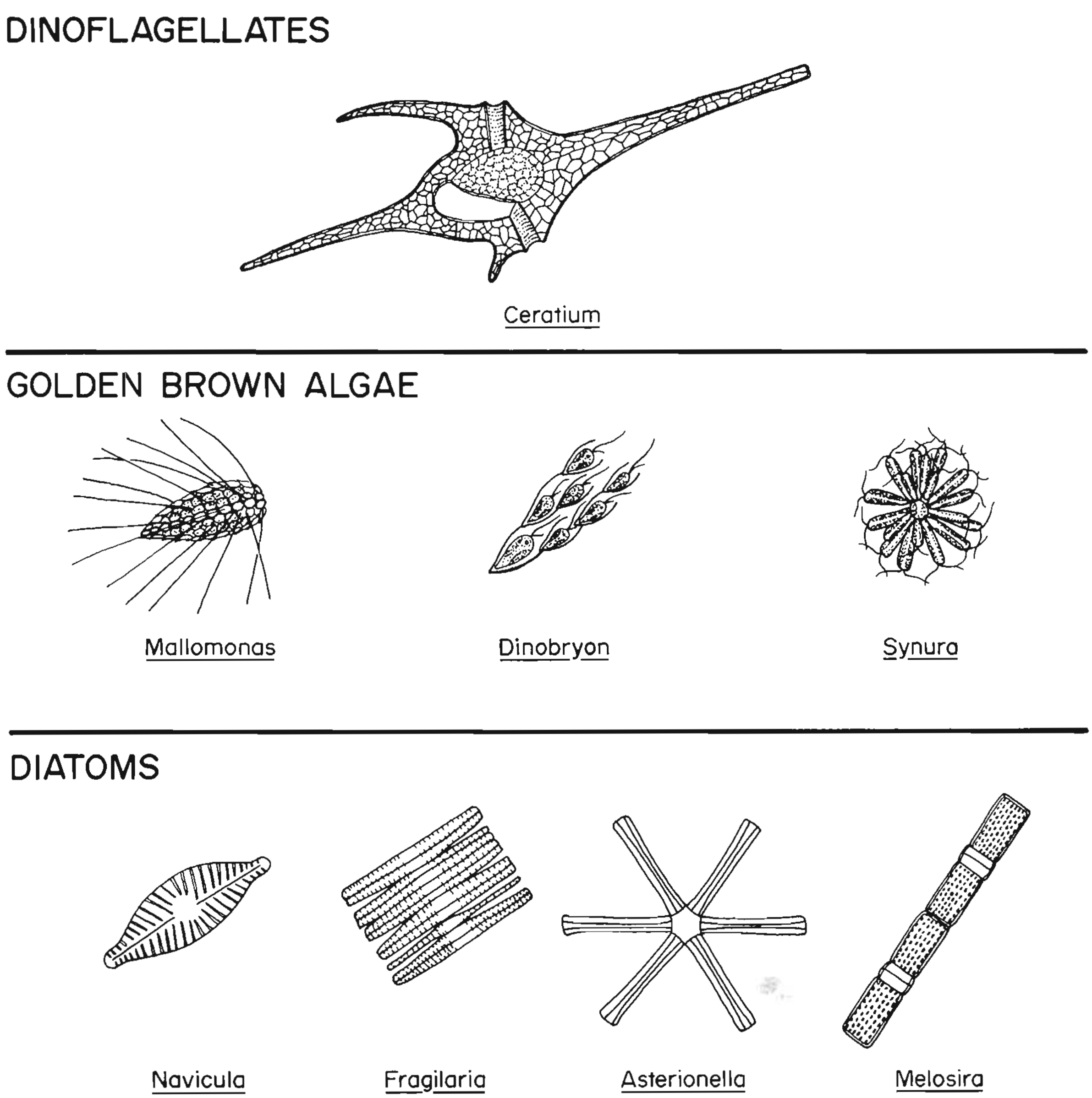

Figure 2. Algae that are primarily golden-brown in color. Unhealthy or dying cells will take on a greenish cast.

The euglenoids that most frequently cause red water are *Euglena sanguinea* and several species of *Trachelomonas* (Figure 1). The color varies from brick red to brown.

Golden-brown Algae

This is a diverse group of algae. It contains three major subdivisions: the yellow-greens, the true golden-browns, and the diatoms. The only yellow-green of aquatic weed interest is *Vaucheria*. It is a mat-forming filamentous alga that appears to be extremely resistant to algicides (more so than *Cladophora* or *Pithophora*). It has become a serious problem in some parts of Europe where heavy diuron (Karmex) treatments have apparently been responsible for selecting resistant strains. It does not seem to be a common problem in the United States although it is present throughout the midwest. *Vaucheria* mats are green in color and may be difficult to identify from green algae. One test is to expose some filaments to iodine. If they turn black, the filaments are members of the green algae; if they just turn a light brown, the filaments are probably *Vaucheria*. The reason for this difference is that green algae store starch which turns blue-black with iodine. The yellow-greens do not contain starch and therefore do not react with iodine.

The golden-browns are microscopic and are either unicellular or colonial. They can bloom in the plankton and are most important as foul taste and odor causing algae. If foul water tastes are a problem, the investigator should look for three possible causes; blue greens (particularly *Aphanizomenon*), golden-browns, and diatoms. The golden-browns will be golden in color, and they do swim. The three most common are *Mallomonas*, *Dinobryon*, and *Synura* Figure 2).

The diatoms are a huge group in terms

of numbers of species and abundance on a worldwide scale. They are both freshwater and marine. They are the dominant phytoplankton in the oceans and provide the base of the marine food chain. Roughly 40% of the world's carbon fixation into organic material is conducted by diatoms! Diatoms can be nuisance organisms, causing blooms (usually brown in color), taste and odor problems, and clogging filters. Some of the most common are *Navicula, Fragilaria, Asterionella,* and *Melosira* (Figure 2). In lakes they tend to bloom primarily in the spring, when the temperatures are cool and plenty of silica is available from runoff (they have silicaceous cell walls). *Asterionella* is a common spring bloomer; *Melosira* a common late summer and fall bloomer. Since they tend not to persist into the summer they are considered less noxious than blue-greens. However, they may persist in cold water lakes where an abundance of silica and other nutrients are available.

Identification Guides to the Algae

There are two fairly inexpensive paperback books on the identification of algae. Both have keys as well as diagrams.

Whitford, L.A. and G.J. Schumacher. 1973. *A Manual of Fresh-Water Algae.* Sparks Press, Raleigh, North Carolina.

Pentecost, A. 1984. *Introduction to Freshwater Algae.* Richmond Publishing Co., Orchard Road, Richmond, Surrey, England. Price is $22.95 and is available from the Mad River Press, Rt. 2, Box 151-B/141 Carter Lane, Eureka, CA 95501.

The Whitford and Schumacher book is much more detailed than Pentecost's because it shows a large number of species of the major genera. One of its big drawbacks is that it does not have pictures of some of the weedy algae such as *Cladophora* and *Pithophora*. It is reasonably good for planktonic algae, however.

Although written to describe the algae of England, Pentecost's book covers all of the important algal genera found in the eastern U.S. It is easier to use than Whitford's book and therefore most people should find it satisfactory. There are some inaccuracies in the keys, but they should be readily caught by the person using them. At the present time this seems to be the best book available.

The other classical algal ID texts such as Smith's *Freshwater Algae of the United States* are out of print. However, Prescott's *Algae of the Western Great Lakes Area* was reprinted in 1982 and is available from Lubrecht and Cramer, R.D.I., Box 244, Forestburgh, NY 12777. The price is $32.50.

Cercospora rodmanii

A BIOLOGICAL CONTROL AGENT FOR WATERHYACINTH

By

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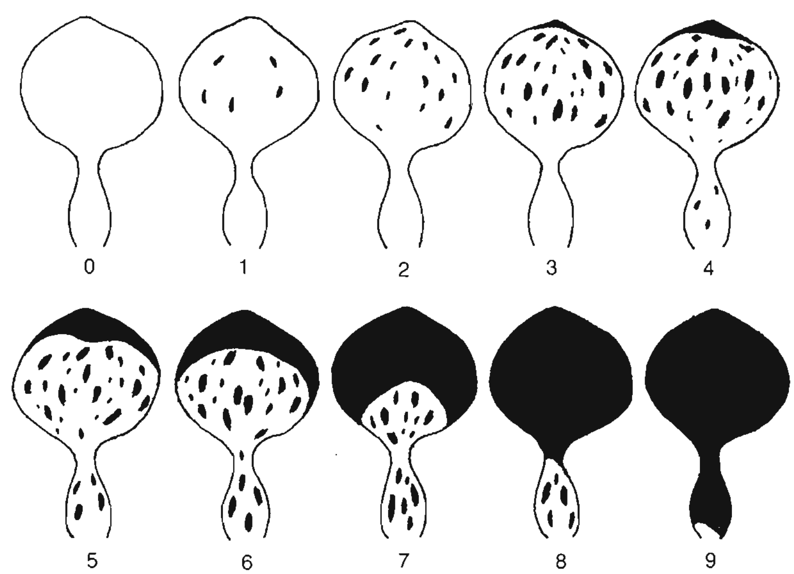
Waterhyacinth, like any other plant, is surrounded in nature by an array of microorganisms, including many virulent pathogens and numerous weak parasites and casual colonizers. Pathogens are the most abundant of the natural enemies of plants and include a variety of agents, such as fungi, bacteria, nematodes, and viruses. In addition to their direct effects in causing diseases, pathogens play an active role in the decline and degradation of plant tissues, leading to a microbiological scavenging and recycling of dead plant tissues. Therefore, it should not be surprising that we often find field populations of waterhyacinths that are covered with dark-brown or bright yellow spots and streaks on leaves and "petioles;" rotting leaf and petiole segments; and decaying roots. These visual signals are expressions of diseases or disease-like maladies caused frequently by simple or a complex of biotic and abiotic factors in which pathogens, parasites, and microbial colonizers commonly play an important role. Pathogens therefore are an important part of waterhyacinth's ecology and hold a tremendous potential as biological control agents.

History of Our Research Program

In 1971, Dr. T. "Ed." Freeman, Dr. F. William Zettler, and this author, working under the auspices of the Plant Pathology Department of the Institute of Food and Agricultural Sciences in Gainesville, undertook a study of diseases affecting aquatic weeds with the aim of developing suitable plant pathogens as biological control agents for waterhyacinth and other serious water weeds. Since very little was known then about pathogens occuring on waterhyacinth and other weeds in Florida and the Southeast, the first research priority was to survey for domestic pathogens. The surveys were however extended to several overseas regions in order to obtain a collection of all known pathogens of waterhyacinth that may have potential as biological control agents. After about three years of domestic and foreign surveys, during which most of the important pathogens of waterhyacinth were collected from around the world and evaluated, a decision was made to concentrate the research efforts on a pathogen native to the United States. This decision was reached partly because of the belief that it would be safer to use a native pathogen that is already in "balance" with our local ecosystem and therefore is less likely than an exotic pathogen to cause any adverse and unexpected effects. Therefore, Dr. Kenneth E. Conway, who was then associated with the project, Freeman, and the author initiated research into the development of *Cercospora rodmanii*, a fungus native to Florida as a biological control agent.

Discovery and Taxonomy

In 1973, what appeared to be a new



Numerical ratings of symptoms: 0—no spots on lamina or petiole; 1—1 to 4 spots on lamina, no petiolar spotting; 2—less than 25 percent of lamina surface with spots, no coalescence or petiolar spotting; 3—less than 50 percent of lamina surface with spots, some coalescence, no petiolar spotting; 4—less than 50 percent of leaf surface with spots, coalescence, some tip dieback, and petiolar spots; 5—less than 50 percent of leaf surface with spots, coalescence, 10 percent tip dieback, and petiolar spotting; 6—less than 75 percent spots, coalescence, 30 percent tip dieback, and petiolar spotting; 7—greater than 75 percent spots, coalescence, 60 percent tip dieback, coalescing spots on petiole; 8—dead lamina, petiole green, but heavily spotted; and 9—dead lamina and petiole (submerged).

Figure 1. A diagrammatic representation of the progression of disease damage caused by Cercospora rodmanii. This diagram is used as a pictorial rating scale in assessing the disease severity.

disease of waterhyacinth was discovered in the Rodman Reservoir, Florida. It was characterized by root rot, leaf spots, and leaf necrosis. At first the root rot was thought to be the primary damage resulting from the disease. However, later the root rot was determined to be a secondary effect, and the leaf spots and the severe leaf necrosis, caused by a *Cercospora* sp., were regarded the primary symptoms. Although a *Cercospora* disease, caused by *Cercospora piaropi* Tharp had been previously found in Florida, because of certain taxonomic and pathologic considerations, the Rodman fungus was treated as a new species (for more on *C. piaropi*, see: Freeman and Charudattan, 1974, Plant Disease Reporter 58: 277-278 and Martyn, 1985, Journal of Aquatic Plant Management 23: 29-32). Consequently, it was justified to create a new species, *C. rodmanii* Conway, for the pathogen from the Rodman Reservoir.

Disease Symptoms

Cercospora rodmanii causes small brown to black spots on the leaves and petioles of the plant. The spots are more numerous towards the tip of the leaf but can occur over the entire leaf surface and the upper portions of the petioles. Because of these spots, the leaf dies from the tip back, with death of tissue gradually spreading towards the base of the leaf until it is killed. The progress of these symptoms is depicted diagrammatically in Figure 1.

Plants with severely infected leaves become "anemic" and stressed. Under severe disease conditions, all leaves on a plant may have dark brown necrotic spots, and the plant may have mostly dead leaves that are water-soaked, causing the plant to sink. In the advanced stages of disease, root deterioration frequently occurs.

Disease stress within a population of waterhyacinth is manifested initially as an overall yellowish appearance of the plants. Numerous severely spotted or dead leaves soon become evident on the plants. As the disease progresses, the entire population of waterhyacinth develops a brownish appearance. At this stage the population begins to decline, and open water appears in areas where previously there had been dense stands of waterhyacinth. As the disease continues to build-up and spread, the mat of vegetation breaks up, and small clusters of heavily diseased plants float away from the mat. The plants in these clusters may have only one to three small yellow leaves with brown spots and numerous dead leaves still attached but sunken beneath the water surface. Finally the entire cluster gradually sinks to the bottom. This type of disease progression, which has been repeatedly observed under controlled conditions, may take several weeks to months and will occur only under severe and sustained disease pressure.

The symptoms described and depicted in Figure 1 are typical and easy to diagnose, but the disease may not be easily recognized in later stages without the benefit of either historical knowledge or early observations. In such cases, it is necessary to examine diseased tissue for the presence of the causal agent with the help of the pathogen's morphology described above.

Epidemiology

In nature, spores produced on diseased tissue are spread by wind and serve to disseminate *C. rodmanii* between locations. Production of spores on diseased tissue is curtailed by temperatures below 13°F but reaches a maximum in the range of 68° to 86°F. The speed and intensity of the epidemic is directly related to the number of spores produced, which in turn is related to the amount of diseased and dead tissue available for sporulation. In a study conducted in 1976, spores were trapped with a spore sampling device near a lake in Gainesville over a 12-month period. It was observed that sporulation reached a peak during the fall and early winter months. High numbers of spores were associated with high disease intensity and low numbers with low disease, suggesting that the number of spores trapped increased as disease intensity increased. As a corollary, the amount of diseased and dead tissues, and hence the pathogen's biotic pressure on waterhyacinth, can be increased by well-timed spray application of formulations containing infective propagules.

Distribution

Since its discovery, *C. rodmanii* has been found on waterhyacinth in areas in Florida other than its original location in the Rodman Reservoir, confirming that it is well distributed in the state. It occurs in several locations along the St. Johns River from Lake Poinsett near Cocoa to Lake George. It is also present in Lake Rousseau on the Withlacoochee River, but not in Crystal River, where *C. piaropi* is predominant. In northcentral Florida, *C. rodmanii* is present in Orange Lake, East Lake Tohopekaliga, and the Suwannee River. It occurs in the irrigation canal systems in Palm Beach and Broward Counties.

Cercospora rodmanii has been confirmed in samples collected from Louisiana at Hayes North, Pecan Island I, Pecan Island II, and Manchac I by the personnel of the U.S. Army Corps of Engineers Waterways Experiment Station. The pathogen has been disseminated on to plants in Lake Concordia in Louisianna and several test locations in Mississippi. The actual distribution of *C. rodmanii* in the Southeast is probably much wider than we have documented so far. However, it is also likely that the occurences of *C. rodmanii* and *C. piaropi* overlap throughout the region, making the field diagnosis of the two species difficult if not impossible.

Efficacy

In terms of vegetative growth, Waterhyacinth is one of the most productive photosynthetic organisms and the biological control efficacy of *C. rodmanii* is related to the growth rate of its host. Under conditions favorable for growth, we have found that waterhyacinth was able to produce one new leaf every 5 to 6 days and thus was capable of outgrowing the disease caused by *C. rodmanii*. We therefore think that when conditions are present that favor disease development and limit leaf production to less than one leaf every 3 weeks, the pathogen could kill leaves faster than the plant could produce new leaves. The plant would then become debilitated and die unless conditions changed to stimulate its regrowth or conditions became less favorable for the disease.

We tested this hypothesis by determining the relationship between the disease caused by *C. rodmanii* and host growth rate at different nutrient levels. This was accomplished by measuring plant growth, disease incidence, and disease severity with the aid of standard techniques. We then calculated the level of disease stress and rate of disease progress required to kill waterhyacinth, there were 20 to 90% reductions in host growth rates (as measured by weekly increments of green leaves) as a result of just one application of *C. rodmanii*. The highest reductions occurred on the lower nutrient levels. However, when the nutrient concentration was at the highest, waterhyacinth grew at a rate faster than the epidemic rate could keep up.

Growth causes a centrifugal spread of waterhyacinth. In the free-floating condition of the plant, new leaves emerge in the center of the plant and older leaves are pushed out and into the water. When the plant is in crowded stands, older leaves are pushed out and towards lower leaves. Apparently, in the process of growth, the host can compensate for diseased and dying leaves with a rapid turnover of leaves. As a result, there was a significantly higher number of dead leaves on diseased plants. This rapid turnover of infected leaves appears to have a sanitational effect by reducing the subsequent spread of the fungus and appears to have been directly related to the observed diminution of the severity of disease from the original to the newer leaves.

During the study, the pathogen killed only the leaves present at the time of inoculation and the fungus on these leaves appeard to be unavailable for subsequent infection cycles due to rapid turnover of leaves. Therefore, long term biocontrol with single applications of *C. rodmanii* is unlikely when the host growth is rapid.

The results therefore confirmed that for practical levels of control of waterhyacinth by *C. rodmanii*, the fungus should be used under conditions that favor low to moderate host growth rates. Other ways of improving the biocontrol efficacy of *C. rodmanii* in the field include: i. using multiple applications of the fungal preparation when waterhyacinth is in the seasonal early growth phase (i.e., late spring in the Southeast) and ii. combining the pathogen with other biotic or abiotic agents capable of retarding waterhyacinth's growth rate. Combinations of the pathogen and insect biocontrol agents or the pathogen and sublethal rates of chemical herbicides are potentially useful approaches.

Safety

Any biological control agent must meet certain safety standards, the most important of which in the case of *C. rodmanii* is its host specificity. It is required that a biological weed control agent be host specific or at least possess an extremely narrow host range. In any event it should be safe against economically or ecologically important nontarget plants. For this reason, the host range of *C. rodmanii* was determined both in the greenhouse and in the field using plants that are taxonomically related to waterhyacinth and those that are economically important to Florida. Eighty-five plants in 22 families were screened but only waterhyacinth was found to be highly susceptible. In some tests, *C. rodmanii* was found on brown spots that developed on dying or older leaves of *Apium graveolens* var. *dulce* (celery), *Cucumis sativus* (cucumber), *Cucurbita pepo* var. *melopepo* (squash), and *Lactuca sativa* (lettuce). In further trials, when reisolates of *Cercospora* obtained from these hosts were inoculated back on the respective hosts and waterhyacinth, only waterhyacinth became infected. Thus, *C. rodmanii* was determined to be a pathogen highly specific to waterhyacinth.

One of the concerns in the use of *C. rodmanii* is whether it will harm fish. To determine this, *Gambusia affinis* (mosquito fish) was exposed to *C. radmanii* in a standard bioassay. At the end of the assay, none of the fish in any of the treatments was adversely affected. In fact, fish in the tank receiving the highest rates fed on the fungus, which was subsequently isolated from their feces. Based on this limited test, it appeared that *C. rodmanii* will pose no threat to fish.

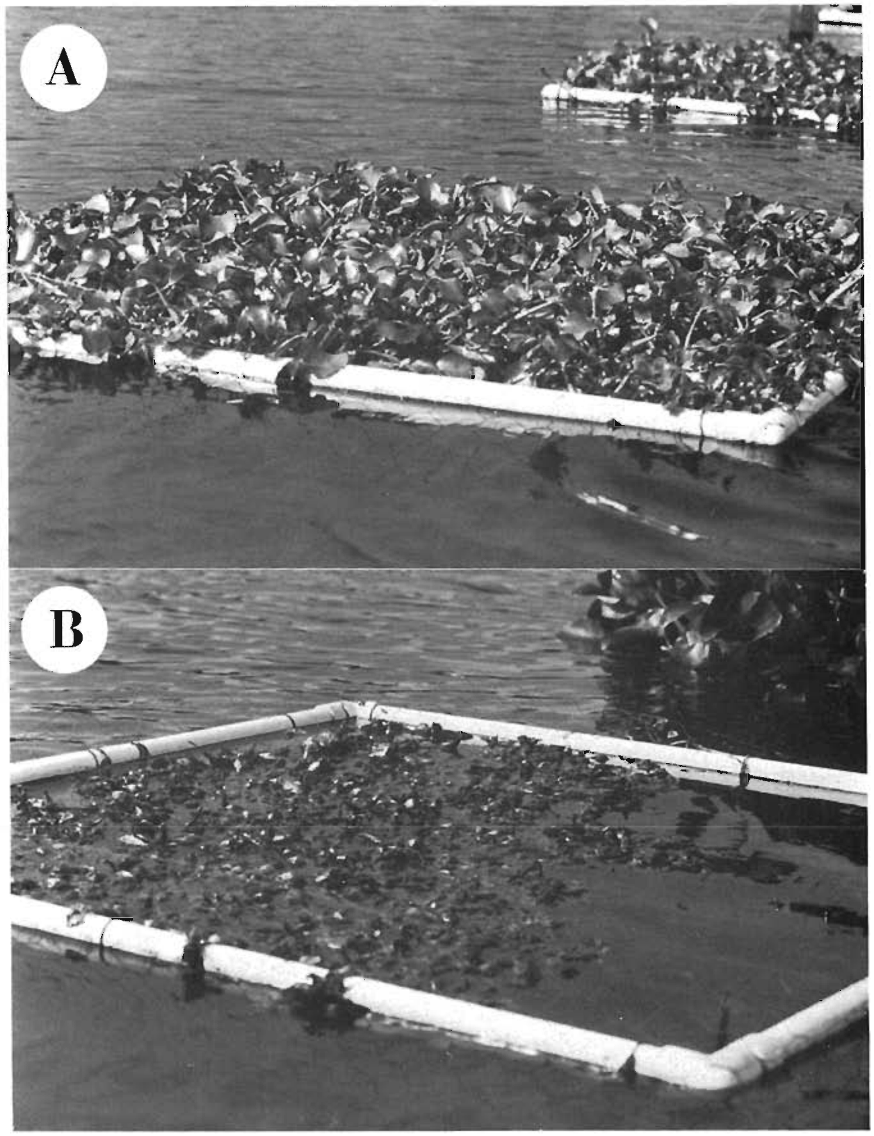


Figure 2. An illustration of the kind of control obtainable with a combination of Cercospora rodmanii and the waterhyacinth weevils (Neochetina spp.) A. Waterhycinth Plants which were subject to season-long stress from the weevils alone. The pathogen was eliminated from this frame by repeated spraying of a fungicide. B. Waterhyacinth plants stressed by both the weevils and C. rodmanii. These frames were part of a controlled study in which an attempt was made to quantify the effects of the weevils, C. rodmanii, and their combinations.

Field Tests

Between 1974 and 1978 *C. rodmanii* was field tested in several locations in Florida and Louisiana using fungus preparations produced in the laboratory. The pathogen was established in test plots usually with just two applications, and the disease typically appeared and began spreading within three weeks, confirming the feasibility of initiating epidemics in nature. The fungus was capable of second spread by wind-borne spores to nonsprayed waterhyacinths in the field.

From these tests it was determined that *C. rodmanii* can severely affect waterhyacinth growth, especially under conditions that favor a reduced growth rate of the plant. Although the greatest effect of *C. rodmanii* was in reducing plant height and biomass, we have found that plant death and the appearance of open water can occur as the result of severe disease pressure.

Attempts at Registration and Commercialization

The use of *C. rodmanii* as a biological control agent for waterhyacinth was patented by the University of Florida, and the University granted a license to

Abbott Laboratories, North Chicago, IL for large-scale production of a standardized preparation of the fungus and commercial development of a microbial herbicide product. Abbott Laboratories developed wettable powder formulations of *C. rodmanii* and obtained a U.S. Environmental Protection Agency Experimental Use Permit (EUP), as required under the biorational Pesticide Guidelines and the FIFRA, to evaluate *C. rodmanii* as a microbial herbicide.

The studies up to this point confirmed *C. rodmanii* to be a potentially successful microbial herbicide. The possibility of registration and commercialization of *C. rodmanii* appeared good. Nonetheless, in 1984 Abbott Laboratories decided not to proceed further with attempts to register *C. rodmanii* on the grounds of technical difficulties in assuring efficacy and economic uncertainties of the marketplace. Currently, we are not actively seeking industrial collaboration, although future collaboration is not ruled out.

Effects of the Combined Actions of Cercospora radmanii and Insect Biocontrol Agents

Until recently, there had been no research aimed at distinguishing and quantifying the effects of insect biological control agents in the absence of the pathogen's effects and vice versa. Even in cases where the effects of insects (for example, *Neochetina spp.*, waterhyacinth weevils) have been well documented, the contribution of the omnipresent microbial agents is usually not determined because of lack of coordinated research efforts. As stated at the beginning of this article, it is my contention that in any population of waterhyacinth in the Southeast that is under severe arthropod attack, pathogens and other microorganisms play an important role in causing premature senescence, browning, cell destruction, and death. Therefore, in 1982 we began a field trial in an attempt to distinguish the biological control effects contributed by arthropods (primarily *Neochetina* spp.) from those of *C. rodmanii*. This was accomplished with the help of insecticide and/or fungicide sprays that minimized the arthropod attacks and/or eliminated the disease as needed according to the experimental design. The test was conducted in moored floating frames (27 sq. ft.) in a lake in Gainesville, and the fungus was applied three times between May and July at the rate of about 0.001 lb. per sq. ft. of the Abbott formulation. At the beginning of the experiment, all plants had *Neochetina* spp. Two insecticides were applied every two weeks: malathion (between May and June) and carbaryl (July and November). Benomyl was the fungicide (also applied every two weeks). Data on shoot height, disease incidence and surface cover were gathered monthly and the experiment was terminated in December just after the first frost.

The combination of *C. rodmanii* and arthropods was capable of eliminating waterhyacinth from the test frames. The fungus alone or the arthropods alone did not completely kill the plants in this study (Figure 2). The fungus had only a slight effect in reducing plant height, but its greatest effect was on leaf browning, debilitation, and death of arthropod-damaged plants. The arthropods alone significantly reduced shoot height (by about 50%), but did not kill the plants. On the other hand, plants threatened with the fungus and arthropods were more severely affected than plants treated with the fungus alone or with the arthropods alone. Plants treated with both *C. rodmanii* and arthropods died six months after the first application of the fungus. Dead plants decayed and sank, leading to open water within the frames. In seven months, the treatments consisting of a combination of *C. rodmanii* and the arthropods yielded 99 percent control (99% surface clearance) of waterhyacinth. Therefore, *C. rodmanii* and the arthropods were compatible and it appears that complete control of waterhyacinth can be obtained in the field by integrating the pathogen and *Neochetina* spp.

Importance of a Microbial Herbicide to the Biological and Integrated Controls of Waterhyacinth

Among pathogens of waterhyacinth, *C. rodmanii* has the greatest potential for practical use. We recommend that it is best used as a microbial herbicide with annual applications of inundative doses of the fungus at a time that is most favorable for infection. In contrast to the microbial herbicide is the classical bio-control agent. In the latter case, a pathogen is introduced into a new region by limited releases of its spores and allowed to reach epidemic levels by its own reproductive capacities. In this process it exerts a weed-controlling effect.

The reasons for recommending the microbial herbicide use of *C. rodmanii* are as follows. Under the conditions in the southeastern United States, *C. rodmanii* has not always produced an economically acceptable level of control naturally without man-induced epidemics. Usually, diseases of weed-controlling intensity occur infrequently in nature. When they occur naturally, the disease incidence and disease severity are often too unpredictable to be of value to biocontrol efforts. Moreover, at certain rapid growth rates of waterhyacinth, it appears that even the combined activities of the arthropods and the natural incidence of *C. rodmanii* may be insufficient to afford control, although the disease that follows arthropod damage is generally the factor that kills the weed. Finally, the principles of plant disease epidemiology teach us that the application of sufficient amount of *C. rodmanii* to waterhyacinth at the beginning of the spring growth, when usually conditions favorable for infection prevail, may result in a disease level of severe, weed-controlling intensity. Therefore, instead of relying on natural occurences of *C. rodmanii*, attempts should be made to disseminate the fungus early in the growing season. This approach, as supported by our results appears capable of yielding economically acceptable levels of control, provided factors that can reduce waterhyacinth growth rate, such as insect biocontrol agents are present on plants.

Summary

Cercospora rodmanii is a virulent fungal pathogen that causes a necrotic leaf spot disease of waterhyacinth. It is a native of Florida and occurs naturally in several locations in the Southeast. It is host specific to waterhyacinth and is a safe biological control agent. Following experimental infections, it was found to cause an extensive damage to the photosynthetic tissues and a gradual debilitation and death of waterhyacinth plants. In field trials with laboratory-produced fungus and industrial formulations, it produced disease epidemics, but its biological control effects varied from a slight reduction in biomass to near elimination of the weed. The latter occurred only when the rate of host growth was already extremely limited by other factors or when other biotic agents were present on the plants at a nonlethal but stressful level. The fungus can be used as a practical microbial herbicide in combination with other biotic and abiotic agents.

Acknowledgements

The author thanks Mr. Thomas Freeman for the original drawing of Figure 1 and the Florida Department of Natural Resources, U.S. Army Waterways Experiment Station, and the Center for Aquatic Plant Research, IFAS for funding of our research.

Sonar: EPA Approved!

By
David P. Tarver

Elanco Products Company
Aquatic Specialist

Sonar (fluridone) a new aquatic herbicide, discovered in 1975 and developed by Eli Lilly and Co., was fully registered by the Environmental Protection Agency on March 28, 1986. After receiving an Experimental Use Permit in 1981, Sonar has been thoroughly evaluated in numerous water systems with a variety of aquatic weed problems. In total, over 9,000 acres of trials were conducted under the 5 year EUP by nearly 200 cooperators. Sonar was tested by water managers in ponds, lakes and reservoirs. Eli Lilly researchers conducted separate trials in canals from 1981-1985. This new registration is the result of extensive laboratory and field testing in order to determine chronic and acute toxicology, environmental fate and herbicidal efficacy.

What exactly is fluridone and what is the mode of action? Fluridone, 1-methyl-3-phenyl-5- -3 (trifluoromethyl) phenyl -4 (IH)-pyridinone, is considered non-volatile; thus, the compound will not evaporate into the atmosphere during or after applying Sonar. Ultra violet light, a component of sunlight, decomposes fluridone into non-herbicidal and non-toxic products. The speed of the photodegradation process is largely governed by the duration and intensity of sunlight, water turbidity and depth of water. Twenty days is the average half-life of fluridone in treated ponds.

In the pH range of 4-14 (which includes most water bodies), 100 percent of the fluridone is in the nonionized molecular form. Therefore, the pH of treated water should have little or no effect on product efficacy. Sonar is not deactivated by adsorbtion from suspended organic or clay particles. This explains its effectiveness in turbid waters.

What is the mode of action of Sonar? Fluridone inhibits the plants ability to produce food. Specifically, carotenoid synthesis is interrrupted. These yellow pigments protect the plants chlorophyll from photodegradation. As carotenoid synthesis is inhibited, the chlorophyll is exposed to photodegradation and is gradually destroyed. The visual symptom of fluridone activity is bleaching or chlorosis on the growing points of the plant.

Susceptible plants will absorb Sonar from the leaves and shoots and from the hydrosoil by way of the roots. It normally takes 30-90 days for control although white tips may be visible as soon as 7 days after treatment. This slow action prevents oxygen depletion and safeguards from potential fish kills.

Acute and chronic toxicological effects have been thoroughly studied. The 96 - hour LC_{50} for fish such as trout, bluegill, catfish and minnows ranges from 7.6 to 22 ppm. This is approximately 76-220 times the normal rate concentration. The 48 - hour EC_{50} values for midge larvae and daphnia are 1.3 and 4.4 ppm, respectively. These values are approximately 10 to 44 times the normal use rate. Results also showed fluridone to have a LD_{50} value of greater than 10,000 mg/kg for mice and rats. Continuous administration of technical fluridone at up to 1,000 ppm in the diet of quail and mallards for six months caused no effects to adults of either species. These included egg shell thickness, hatchability and survival of the young. These fluridone concentrations are between 10,000 and 50,000 times that found in fluridone treated water immediately after treatment. Based on extensive testing it has been concluded that when used as directed, Fluridone does not pose a risk as an acute poison or as a chronic toxicant. Sonar has received a "caution" designation as a result of this low order of toxicity.

Does Sonar control all aquatic plants? No, Sonar is not a sterilant and in fact is a very selective aquatic herbicide. Many submersed vascular species are susceptible when treated just prior to initiation of weed growth or during periods of active growth. These species include coontail, milfoil, hydrilla, egeria, bladderwort, naiad, pondweeds and others. Control of susceptible species normally last from 12-18 months. Sonar does not control algae. However, due to slow action of Sonar, algal blooms have rarely occurred following a treatment.

Some emergent plants such as spatterdock, water lily, and paragrass are susceptible. Duckweed and water-meal are floating species controlled by Sonar. All susceptible species listed in "Sonar Guide to Water Management" are best controlled when treatment is made just prior to or during periods of rapid growth. Sonar is a systemic herbicide and rapid growth enhances product uptake and efficacy. Treatment during dormancy normally minimizes or eliminates herbicidal effects.

Adding to Sonar's selectivity characteristics is a list of tolerant aquatic species. Some tolerant plants include knotgrass, pickerelweed, pennywort, bullrush, waterlettuce, chara, nitella, bacopa, water hyacinth and planktonic algae. Other species are considered intermediate because only partial control may occur. These are species such as tapegrass, torpedograss, Illinois pondweed, slender spikerush and cattail. This classification of susceptible, intermediate and tolerant places aquatic species in broad categories; however, a wide margin of selectivity is available with Sonar depending upon the timing of treatment rate and formulation selected. A management objective should dictate the treatment strategy.

Water managers typically manage water systems for multi-purpose uses. Obviously, certain waters serve a critical role, such as a potable water supply, and should be managed for select uses. Each water resource should receive a management plan oriented toward meeting all the needs of that particular water.

Most federal and state conservation agencies agree that some native aquatic vegetation is desirable in public waters. Aquatic plants serve a useful role in providing food and cover for invertebrates, fish and wildlife. However, an over abundance of plants, native or exotic, is detrimental to the aquatic ecosystem. Detrital buildup decreases fish spawning areas and lowers water pH. Aquatic plants also provide breeding grounds for mosquitoes and restrict water flow in canals and rivers. Numerous other negative impacts of an over population of aquatic plants are well documented.

How does Sonar fit into an aquatic plant management program for lakes, ponds, canals and rivers? Once a managment objective is established how can Sonar be used to obtain the desired results? Again herbicidal selectivity is imperative if non-target species play a role in the management plan.

Three formulations of Sonar are now available for use in all water sites except rivers. Only the SRP (Slow Release Pellet) formulation is approved for river use. This pellet slowly releases fluridone over a period of 7-14 days. It is recommended that large areas in slow moving rivers and canals be treated. Rapidly flowing sites should be treated during periods of minimum flow, optimumly in the spring. Poor product efficacy is likely if application is made during rapid flow. Water retention in canals will improve efficacy.

All formulations are approved for use in ponds, lakes, reservoirs and canals. Herbicidal efficacy is improved by using Sonar A.S. for control of several species such as duckweed and water lilies. For control of these species in static waters a spreader/sinking agent is recommended. Use the recommended Sonar label rates which vary with average water depth. Optimum control is provided when the entire pond is treated during periods of active duckweed growth. Spring and summer treatments of various water lily species provide optimum control. A surface spray of emergent water lily has proven effective. Compete coverage is not required when treating ponds or large areas — but recommended.

Water lilies (*Nuphar, Nymphaea* and *Nelumbo*) are often non-target species growing in association with submersed target species. A different management plan and treatment method is required in order to selectively control the target species. Field data indicates a very early spring (January-March in Florida) treatment with one of the pellet formulations or A.S. and a polymer will selectively control submersed weeds such as highly susceptible *Hydrilla* and *Cabomba* yet minimize impact on emergents. This response basically holds true for other growing submersed species and native emergent plants treated during dormancy. Likewise, *Vallisneria* and *Potamogeton illinoensis* typically increase in abundance following a *Hydrilla* spring or fall treatment. These species are only controlled by Sonar at highest label rates or by total pond treatments. Long term control of *Hydrilla*, varying from 12-20 months, is well documented. this long term control is partially a result of tuber control afforded by Sonar. Early spring and fall treatments provide optimum control results; however, summer treatments are also effective yet require a significantly longer period for plant drop out. This slow decay process does not severely stress dissolved oxygen levels thus protecting against potential fish kills.

The Sonar label requires that no application be made within one-fourth mile (1320 ft.) of any potable water intake. The EPA has determined that when Sonar is applied according to label directions, fluridone concentrations will not exceed 0.15 ppm. Treated aquatic sites may be used immediately for swimming, fishing and domestic uses.

Sonar A.S., like most conventional aquatic herbicides, can be applied with any type of spray equipment. Hand guns, booms, trailing hoses and aerial booms may be used. A polymer mixture of .25% to .50% is normally recommended. A total of 25 to 100 gallons mix per acre is standard. Sonar does not control algal species and use of an algicide with Sonar has proven effective when algae is present on target vascular species. Depending upon water pH and hardness, copper sulfate or chelated copper is recommended. Follow label rates on all herbicides.

Any pellet or fertilizer spreader can be utilized when applying Sonar 5P or SRP. Seymour and Cyclone spreaders and pellet blowers are commonly used. Swath width is normally 25 ft. and only 3-6 minutes are required to treat an acre. Sonar may also be aerially applied through granular spreaders.

In summary, a new generation of aquatic herbicide is available as a result of over ten years of developmental efforts.

Surface Area of Aquatic Macrophytes

By

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Department of Fisheries and Aquaculture

Aquatic macrophytes are important to lake and river systems. When the study of the plant itself is the goal, weight per area or areal coverage are the most common measurement used to characterize the plant. When the plant is considered a substrate for the growth of other plant and animal life, however, surface area of the plant becomes the most important parameter.

Examining aquatic macrophytes as a substrate investigators (Dvorak and Best, 1982; Schramm, et al 1984) found more epiphytic algae (periphyton) and invertebrates per weight of plant, on submersed than emergent aquatic macrophytes. They hypothesized that this was due to a greater surface area to biomass ratio on submersed than emersed plants. Thus the objective of this study was to quantify the surface area to biomass ratio on several submersed and emergent aquatic macrophytes.

Harrod and Hall (1962) developed a method in which the surface area of a plant was determined by the weight of a thin layer of detergent covering the plant. We modified this method through the use of a non-ionic, oil concentrate adjuvant (KAMMO)[1] and a calibration curve of sufactant weight versus known, calculated surface areas. Surface areas were measured for the following plants (collected from the Little Wekiva River) by tracing each plant part (only underwater parts) and measuring their area with a planimeter: *Nuphar luteum*, *Hydrilla verticillata*, *Brachiaria purpurascens*, *Vallisneria americana*, *Ceratophyllum demersum*, and *Chara* spp. The calculated regression line, based on the surfactant weight necessary to form a thin film on a minimum of four samples of each plant type versus the measured surface area was:

(1) Log Surface area (cm^2) $= 1.72 + 0.44$ Log Surfactant weight (gm)
Total number of samples = 34
Coefficient of Determination (R^2) = 0.86

Plant samples were then collected from eight rivers (Table 1) between August and December 1985. The weight of surfactant on each plant sample was used with equation one to determine the surface area of the sample. The plant was then dried at 60°C for 24 hours and weighed. Surface area to biomass ratios were meaned by plant species and recorded as cm^2 per gm. dry weight of plant (Table 2).

The mean surface area to biomass ratio for submersed and emergent plants were 319 and 247 cm^2 per gm. dry wt., respectively (Table 2). These data may explain why more periphyton and invertebrates are found on equal weights of submersed than emergent plants. When aquatic macrophytes are studied as a substrate, structure or refuge for other organisms it would be advisable to measure surface area instead of biomass or areal coverage.

TABLE 1.
Study of rivers and counties.

Rivers	County
Little Wekiva	Seminole
Alexander Springs	Lake
Ichetucknee	Columbia
Aligator Creek	Bradford
Rock Springs	Orange
Little Econlockhatchee	Orange-Seminole
Wacissa	Jefferson
Big Wekiva	Orange-Seminole

[1] Trade name of Helena Chemical Company

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Table 2. Mean surface area to biomass ratios (cm^2/gm. dry wt.) for 7 submersed and 11 emersed aquatic macrophytes collected from 8 different Florida rivers. Grand means are recorded ± 95 percent confidency intervals.

Plants	Number of Samples	Surface area to biomass ratio (cm^2/gm. dry wt.)
Submersed Plants:		
Eleocharis baldwinii	5	264
Sagittaria kurziana	13	309
Egeria densa	9	229
Hydrilla verticillata	30	535
Vallisneria americana	67	233
Najas quadalupensis	10	375
Potamogeton pectinatus	2	207
Grand Mean	**136**	**319 ± 45**
Emersed Plants:		
Sagittaria lancifolia	1	165
Alternanthera philoxeroides	7	206
Limnobium spongia	1	394
Nuphar luteum	30	270
Pontederia lanceolata	6	215
Typha spp	3	197
Hydrocotyle umbellata	14	374
Colocasia esculenta	6	225
Brachiaria purpurascens	21	166
Zizania aquatica	3	249
Micranthemum umbrosum	1	356
Grand Mean	**93**	**247 ± 51**

Native Insect Enemies of Aquatic Macrophytes Beetles

by
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Kim H. Haag[2]
Dale H. Habeck[2]

Introduction

This is the second of three articles discussing native insects that attack aquatic macrophytes in Florida. The moths, or Lepidoptera, were discussed in the first article. The beetles, or Coleoptera, which is the largest insect order, will be discussed here. Beetles comprise about 30% of all known animal species, and there are almost as many beetle species (ca. 300,000) as there are plant species. Thus, it is not surprising that more species of beetles attack aquatic and wetland macrophytes than do species of any other insect order. Beetles can be recognized by their hard forewings, or elytra, which are modified to cover the membranous hindwings.

Unlike other important insect orders that attack aquatic macrophytes only when the insects are larvae (moths, flies, and caddisflies), both beetles and their larvae are herbivores. Often, however, beetles feed on a larger variety of plant species than do their larvae. Unfortunately, the larval host plants and the life histories are unknown for many of the species that feed on macrophytes.

Beetles attack not only emergent macrophytes but also floating and submersed species. Because of the wide range of host plants, beetles have developed a variety of physical and behavioral adaptations for the aquatic habitat. Many species have hydrophobic body surfaces, or integuments, which can not be wetted. There bodies have become surrounded by air as they enter the water. They are able to remain underwater from several hours to several days before the oxygen in the air layer is exhausted. Some species have perfected this adaptation by the formation of a platron, which is a thin, very tightly held layer of air surrounding the body. This air is held by special hairs, scales, or cuticular projections. The insect does not exhaust the oxygen in the plastron because as oxygen is used, new oxygen diffuses from the water to replenish the supply. These plastron bearing insects can remain submerged almost indefinitely if the water has a reasonable supply of oxygen.

Another way to obtain oxygen while underwater is to tap a plant's air supply. This is the most common method for beetle larvae and pupae (the resting stage). They remain inside the plant, or some pupae rest in cocoons that are attached to the plant. Air escapes into the cocoon from wounds made in the plant by the larva prior to making the cocoon. Larvae of some beetles have hooks that they insert into the plant to obtain air.

Beetles with plastrons swim beneath the water surface, but many species swim only on the surface. Others apparently cannot swim, but move slowly and poorly on the surface as if they are trying to walk. These species usually enter the water by crawling on a plant. Some small species walk on top of the water surface, and a few species walk along the underside of the surface submerged in the water and upside down.

Apparently, most if not all, aquatic plant feeding beetles are able to fly. Species that feed on widely scattered hosts, for example waterlily leaves or watermilfoil flowers, may need to be able to fly at all times unless they are good swimmers. Other species, however, that feed on closely spaced plants, for example, waterhyacinth, azolla, rice, and hydrilla, do not need to fly again immediately once they have located a host plant population. The wing muscle of these species decrease in size after migration and their energy reserves and body space are utilized to produce eggs. Wing muscles are formed at the expense of eggs when it is time to migrate.

The natural enemies of aquatic plant feeding beetles have not been well studied. Certainly, vertebrate predators, such as birds, fish, and frogs, as well as invertebrates like spiders and predceous insects, feed on beetles. A white fungus, *Beauveria bassiana*, is a common disease of aquatic beetles in Florida, but we have not observed epidemics. Various other fungi have also been recorded. Protozoans, which are generally debilitating rather than lethal, have been found in some aquatic beetles. Parasitic wasps have also been reported from some aquatic beetles, and undoubtedly more examples will be found when the life histories of more beetles are studied. Unfortunately, the impact of natural enemies on their host populations can not be estimated at present.

The beetle species that we discuss have been chosen either because they are among the most common and noticeable species or because of their special interest to us. The following examples should provide a good foundation for understanding and appreciating this important group of insects.

Family Chrysomelidae

The chrysomelids are commonly called leaf beetles because they generally feed on leaves. The Colorado potato beetle, asparagus beetles, cucumber beetles, and various flea beetles are well known crop pests in this family. Two aquatic leaf beetles that are often noticed by aquatic plant managers are the waterlily leaf beetle, *Pyrrhalta nymphaeae*, which was featured in two recent *Aquatics* articles, and the imported alligatorweed flea beetle, *Agasicles hygrophila*. Flea beetles deserve their name because they have large hindlegs and jump like a flea.

Two native flea beetles, *Disonycha glabrata* and *D. pennsylvanica*, are often mistaken for their close relative, the alligatorweed flea beetle. This mistaken identity would not be of much importance except that the native species feed on smartweeds and amaranths in addition to alligatorweed. Occasionally we have been told that alligatorweed flea beetle adults have switched hosts only to find that a sharp-eyed observer has found one of these native species on its native host plants. The natives have an orange pronotum (the area between the head and wing covers) compared to a black pronotum in the alligatorweed flea beetle. Both larvae and adults of *Disonycha* eat holes in leaves. The pupa is found in the soil on shore. About 10 species of *Disonycha*, most of which feed on terrestrial plants, are found in Florida.

A flea beetle that is quite noticeable when abundant is *Lysathia ludoviciana*.

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This bluish-black species attacks both floating waterprimrose and parrotfeather. All stages have been found on the leaves of parrotfeather although only adults have been reported so far on waterprimrose in Florida. Larvae have been reported on creeping waterprimrose in Texas.

The damage of an azolla flea beetle, *Pseudolampsis guttata*, is much more noticeable than the beetle itself, although even its damage is overlooked by most observers. This tiny (2.5mm) gold and silver beetle is beautifully camouflaged when sitting on azolla. The eggs are laid on the underside of the plant, but the larvae feed only on the emersed leaves. The larvae appear black until fully grown, when it becomes apparent that they are actually green and covered with minute black plates. The light brown, parchment-like cocoon sitting on the azolla is actually the most recognizable sign that this species is present. This flea beetle, alone or together with a weevil, *Stenopelmus rufinasus*, can destroy azolla mats. Typically, the insect population builds to enormous numbers before the damage is noticeable, then very quickly the mat turns brown or grayish as most of the emersed leaves are eaten. The damaged plants sink and the few surviving plants begin the cycle again.

The long-horned leaf beetles, *Donacia* spp., are the most common aquatic chrysomelids. They are elongated with relatively long antennae and are larger than most of their relatives (5-15mm). Often they are metallic bronze, green or blue in coloration. These beetles are common on the flowers or leaves of waterlilies, spatterdock, and watershield. They fly at the slightest disturbance. About 10 species occur in Florida and a variety of life histories are represented. Generally they lay their eggs in groups on the undersides of floating leaves or on submersed leaves. The egg masses on waterlily are placed along the margins of holes eaten in the leaves by the females and are covered with an opaque, whitish, gelatinous material. The white grub-like larvae of most species apparently feed on the roots or lower stems. A close examination of a larva reveals two small dark spines near the tail. These spines, which are characteristic for this group and its close relatives, are reportedly used to obtain oxygen from the plant. Larvae that have not yet changed to pupae can often be observed in the elongate parchment-like cocoons attached to uprooted floating rhizomes of spatterdock. Adults breathe, when submerged, from plastron-like air bubbles around their bodies and some species apparently have true plastrons. Unfortunately, we do not know what role this important group of beetles plays in the regulation of our native plant populations in Florida.

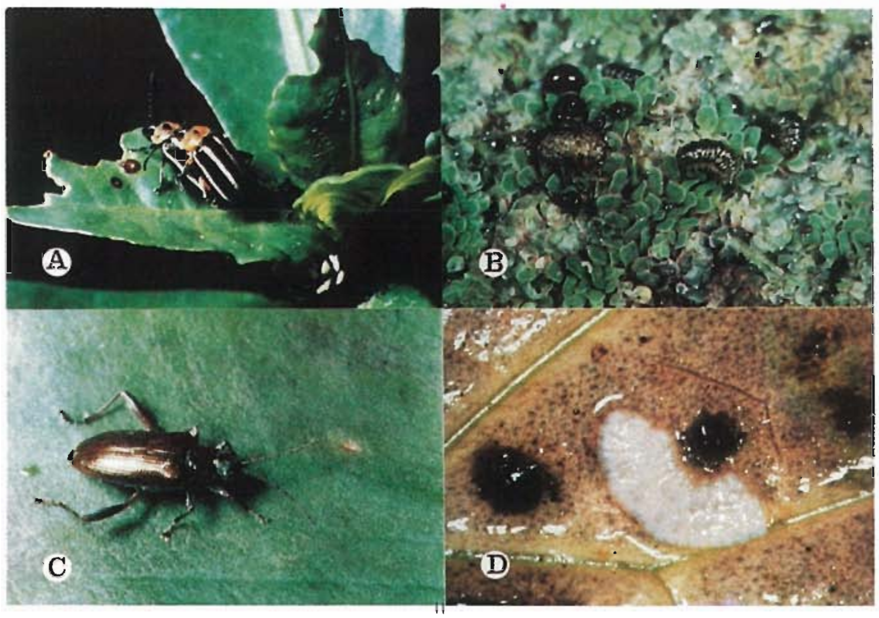

Figure 1. A. Disonycha glabrata on alligatorweed, B. Cocoon and larva of azolla flea beetle, C. Long-horned leaf beetle, D. Egg mass of long-horned leaf beetle.

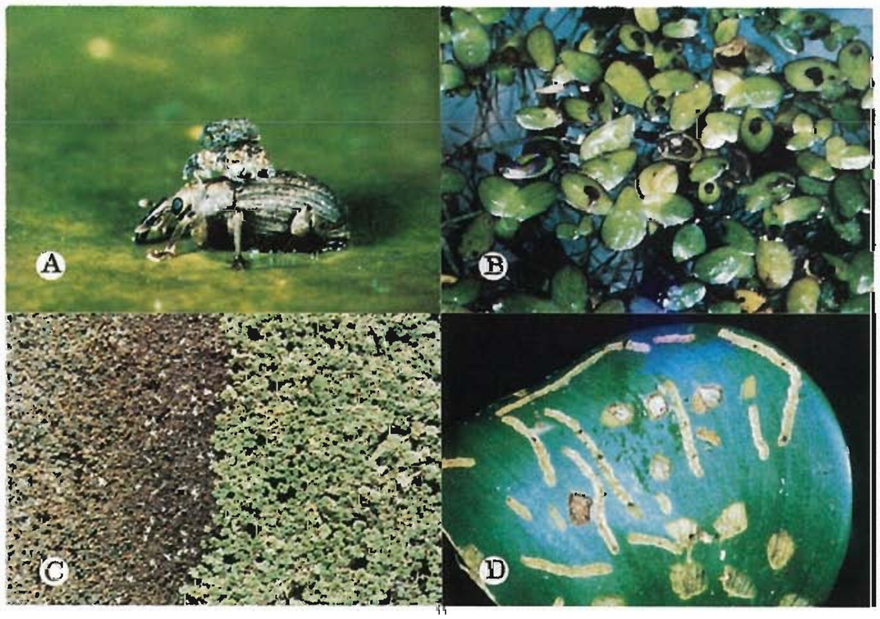

Figure 2. A. Duckweed weevil (top), azolla weevil (center), waterhyacinth weevil (bottom), B. Larva (center) and damage of duckweed weevil, C. Azolla damaged (left) by azolla weevil, D. Feeding scars of Onychylis (narrow) and waterhyacinth weevil (broad) on waterhyacinth.

Family Curculionidae

Weevils, or curculionids, are characterized by elongated snouts. The mouthparts are at the tip of the snout. The cotton boll weevil, alfalfa weevil, and citrus weevil are crop pests in this family. The introduced waterhyacinth weevils are the aquatic species most familiar to aquatic plant managers. Probably more species of weevils feed on aquatic plant managers. Probably more species of weevils feed on aquatic macrophytes in Florida than species in any other family of insects.

The genus *Bagous*, which is the group of greatest interest to us in our program for biological control of hydrilla, is well represented in Florida by native species. About 17 species occur in our state and the larval host plants are still unknown for many of them. Adults of at least some species have plastrons. Larvae tunnel inside plant tissue, and the dark serpentine mines of a common and abundant species, *Bagous lunatoides*, can be readily seen in the floating leaves of frogbit. Larvae also mine in the frogbit stolons and leaf petioles where they pupate.

The rice water weevils, *Lissorhoptrus oryzophilus* and *L. simplex*, are closely related to *Bagous*. Long hairs on the legs of the adults aid them when swimming underwater much like rubber fins aid us. They are major pests of rice, but they also attack other aquatic grasses. The larvae attach to roots by a row or dorsal hooks through which they obtain oxygen. In the autumn, many adults fly to wooded areas to overwinter in the leaf litter, under bark, or in Spanish moss.

Four interesting species related to the preceding are *Stenopelmus rufinasus*, which attacks azolla as mentioned earlier; *Cyrtobagous salviniae*, a South American immigrant now found throughout Florida which attacks salvinia; *Neohydronomus pulchellus*, a South American native currently being evaluated in quarantine by one of us (D.H.H.) for control of waterlettuce; and *Onychylis nigrirostris*, which attacks pickerelweed. The adults of this latter species, which are about half the size of a waterhyacinth weevil, make feeding scars on the leaves of both pickerelweed and waterhyacinth. These scars are narrower and longer than those of the waterhyacinth weevils. Larvae tunnel in the pickerelweed stems. The cocoons of *S. rufinasus* are black which easily distinguishes them from those of the azolla flea beetle. The yellow to red larvae are usually covered with a droplet of black liquid excrement.

Duckweed and giant duckweed leaves often look like someone has attacked them with a tiny paper punch. The circular holes are made by the adults of our smallest (1-1.5mm) aquatic weevil, *Tanysphyrus lemnae*. The yellow larvae mine inside the leaves and only a translucent shell remains when they move to a new leaf. We have found pupae inside leaves, although they are also reportedly found on shore. This species ranges throughout the U.S. to South America and from Europe to Japan. Although they often damage many leaves, destruction of entire mats has not been reported and we have not seen damage comparable to that observed on azolla mats for the azolla beetles.

Arrowheads are reported hosts of many insect species. If the flower stalks of a large arrowhead, *Sagittaria gramineae*, are split open when globules of dried white latex are found on them, all stages of *Listronotus* weevils may be found. The latex covers openings the larvae make to the outside. The adults found inside the stalk are soft and newly formed and rest there while hardening. Later they can be found feeding on the fruiting heads.

A group of small, compact weevils, the ceutorhynchines (suit-o-rin-kines), have adapted to the emersed portions of macrophytes. There are a few species that are as well adapted to submersed macrophytes as are the *Bagous* weevils, but those species are not found in Florida. Adults of *Parenthis vestitus* feed on the flowers and flower stalks of Eurasian and other watermilfoils. They fly readily when disturbed and they swim well on the surface of the water, but not underwater. The yellowish larvae feed initially in the flower buds, then bore into the stalk. They tunnel downward into the stem where they make brown, spherical, parchment-like cocoons. Other ceutorhynchine species make similar cocoons but externally on the leaves of the host plants. These cocoons are usually easily spotted when partially eaten leaves or leaves with irregular shaped holes are searched for the culprits who damaged them. The larvae, on the other hand, are often difficult to spot because they are covered with a viscous dark colored substance, believed to be excrement, and appear to the uninitiated observer as merely droplets of some unpleasant looking liquid. Some of these other ceutorhynchine genera are *Perigaster* on some waterprimroses, *Rhinoncus* on docks and smartweeds, and *Pelenomus* on parrotfeather, docks, and waterprimroses.

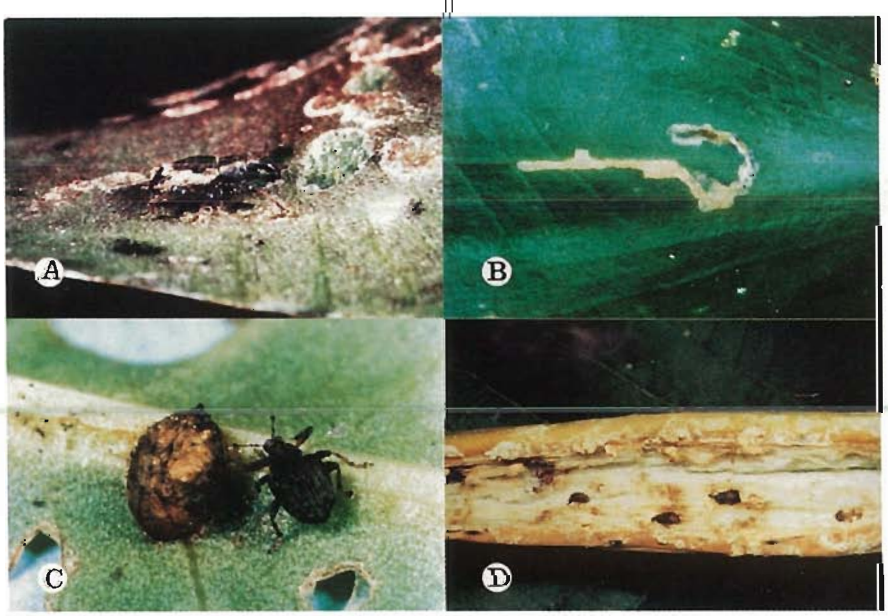

Figure 3. A. Camouflaged Bagous lunatoides on frogbit, B. Larval tunnels of B. Lunatoides in frogbit, C. Cocoon and adult Pelenomus on dock, D. Listronotus adults in arrowhead flower stalk.

Other Families

Only a few species in other families of beetles, for example Hydrophilidae, Haliplidae, Dryopidae, and Helodidae, have been reported to feed on aquatic macrophytes. Little is know about their development, there feeding relationships, or their effects on the plants. Most are small and are not usually encountered. Adults of one species in the family Scarabaeidae, the May beetles or June bugs, are occasionally found burrowing into crowns of waterhyacinth. This species, *Dyscinetus morator*, is black or dark brown and about 1.5-2cm long. The terrestrial larvae feed on the roots of grasses and other plants.

In Conclusion

Much has been written about some introduced leaf beetles and weevils as biocontrol agents of aquatic weeds. Equally interesting, and perhaps equally important, are the native beetles that attack aquatic plants. A thorough understanding of their roles in plant regulation would undoubtedly aid future efforts to manage aquatic systems and should be a goal of aquatic scientists.

Acknowledgements

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They do not bioaccumulate in the food chain, nor do they bind or leave residues in the hydrosoil. And Pennwalt's aquatic herbicides provide an ample safety margin to fish, shellfish, birds, and other wildlife.

For a complete aquatic weed and algae control program, Pennwalt offers four choices:

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Special Notice

Florida Aquatic Plant Management Society Scholarship and Research Foundation, Inc.

On August 19, 1984, aquatic plant management lost a valuable asset and friend with the passing of Bill Maier. Those who knew Bill understood his love of the outdoors and his understanding of the role of professional aquatic plant managers and dedicated research scientists. It was only fitting that Bill's service to the profession be honored in a manner in which he truly believed, plant management. Thus the concept of a William L. Maier Memorial Scholarship was pursued and the Florida Aquatic Plant Management Society Scholarship and Research Foundation was formed and established in 1985 as a separate corporation. The first annual scholarship was awarded to Ms. Patricia L. Smith at the 1985 annual FAPMS meeting.

To date most of the contributions to the Foundation have been either from Bill's immediate family or the Board of Directors of both APMS and FAPMS. However, many individuals have expressed a desire to contribute to the fund once the Foundation received its tax exempt status. The FAPMS Scholarship and Research Foundation, Inc. was finally granted tax exempt status by the Internal Revenue Service under the provisions of section 501(c)(3) of the Internal Revenue Code. According to IRS "Donors may deduct contributions to the Foundation. Bequests, legacies, devises, transfers, or gifts are deductible for Federal estate and gift tax purposes."

The immediate goal of the Foundation is to obtain enough capital that the interest on the investments will support the establishment of several annual scholarships in the range of $300 to $500. At present only one such award is possible and applications have been solicited for the 1986 scholarship to be awarded at the FAPMS annual meeting in October, 1986. Those of you who would like to honor Bill's memory and commitment to aquatic plant management through a contribution can do so by sending a check to:

The Florida Aquatic Plant Management Society Scholarship and Research Foundation, Inc.
c/o Dr. Joseph C. Joyce Sec/Tres
7609 SW 19th Place
Gainesville, Florida 32607
(904) 376-0732

All donors will receive a letter of acknowledgement of the contribution and a copy of the letter of tax exempt status from IRS.

Your contribution in Bill's memory will be most appreciated.

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Footnotes

1. Research Entomologist, ARS, USDA, P.O. Box 1269, Gainesville, Florida 32602.
2. Research Associate and Professor, respectively, Dept. Entomology and Nematology, Univ. of Florida, Gainesville, Florida 32611.

AQUA VINE

MOVING UP

Recent personnel changes at the St. Johns River Water Management District have shifted responsibilities around, moving Wayne Corbin up the ladder to their Aquatic Weed Supervisor. Wayne oversees aquatic weed control activities at Palatka and Melbourne field stations. Although the mahogany boat at the office will consume more of Wayne's available work time, he will also spend much of his time on an airboat assisting in weed control activities.

DNR

Michigan State Graduate Rob Kipker has recently joined the research staff at the Bureau as a full time Biological Scientist. Rob has been with the Department for several years on a temporary basis working with bioassay and fish culture techniques.

SFWMD

The new Assistant Weed Management Coordinator for the South Florida Water Management District is Mr. Ed Terczak. Ed brings to the aquatic weed management section 15 years of experience with the District's Environmental Section. Ed's familiarity with the pesticide industry comes from his mosquito and aquatic weed control experience he gained prior to coming to the District.

CAW

As a result of the newly created Department of Fisheries and Aquaculture being located at the Center for Aquatic Weeds, many personnel changes have recently occured.

President Emeritus and Professor of Medicine, Robert Q. Marston, M.D. comes to the Department as a part-time faculty member utilizing his medical expertise in the grass carp physiology research project.

Fisheries geneticist, Donald Campton, comes to the Department from Davis, CA, where he received his Ph.D. Don spent 5 years in the state of Washington as a fish biologist and plans on studying population genetics of freshwater and marine fisheries, along with participating on the grass carp research project.

IN MEMORIAM

On December 26, 1985, Alex Fears, Jr. was killed in a hunting accident on private land adjacent to Chattahoochee State Park in Houston County, Alabama. While unloading lumber from the bed of his pickup truck, a sheet of plywood struck a loaded double-barrel shotgun which discharged and killed Alex immediately.

A lifetime resident of Greenwood, Florida, he worked for the Northwest Florida Water Management District (NWFWMD) for five years as an Aquatic Plant Control Technician. Prior to his employment with the District, he worked for the Production Credit Association for five years. He was a diligent and dependable worker, and will be missed by all of those who knew him.

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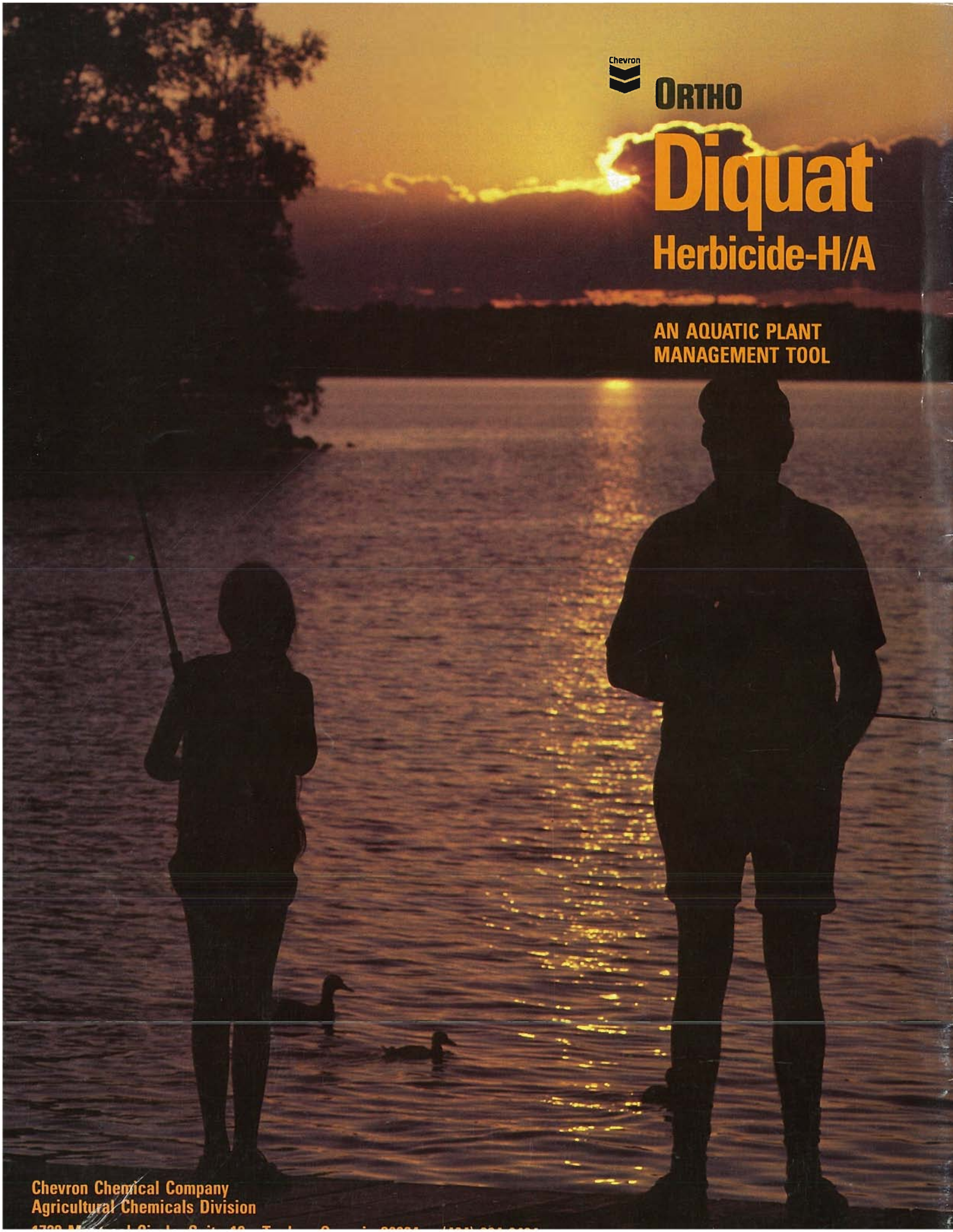
water quality projects — with DER dollars.

At the time of this printing, new legislation is proposing an additional 1.3 million dollars to be taken from DNR this year. Add this with the $737,000 unrequested transfer to the Game and Freshwater Fish Commission plus the 1.5 million taken in April equals a grand total of 3.537 million dollars! That amount would have kept Florida's waterways free from floating plant problems for nearly two years! Without this money, an overabundance of weeds will again dominate Florida's waters. DNR's Aquatic Plant Control Funds will no longer be sufficient to meet the needs of our most valuable natural resource — water.

What can you do to impact this situation? Be visible and squeak! Let your legislators know you do not support money being taken from the aquatic plant trust fund to finance the Lake Apopka Project. These funds are needed for its original intent.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