A

APPENDIX A

Public Participation Materials

Presentation Outline

- Onterra, LLC
- Why Create a Management Plan?
- Elements of a Lake Management Planning Project
  - Data & Information
  - Planning Process

Onterra LLC Lake Management Planning

Onterra, LLC

- Founded in 2005
- Staff
  - Three full-time ecologists
  - One part-time paleoecologist
  - Four full-time field technicians
  - Four summer interns
- Services
  - Science and planning
- Philosophy
  - Promote realistic planning
  - Assist, not direct

Onterra LLC Lake Management Planning

Why create a lake management plan?

- Preserve/restore ecological function to ensure cultural services
- To create a better understanding of lake's positive and negative attributes.
- To discover ways to minimize the negative attributes and maximize the positive attributes.
- Snapshot of lake's current status or health.
- Foster realistic expectations and dispel any misconceptions.

A goal without a plan is just a wish!

Onterra LLC Lake Management Planning

Elements of an Effective Lake Management Planning Project

Data and Information Gathering

Environmental & Sociological

Planning Process

Brings it all together

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Data and Information Gathering

- **Study Components**
  - Water Quality Analysis
  - Watershed Assessment
  - Paleocore Collection & Analysis
  - Aquatic Plant Surveys
  - Fisheries Data Integration
  - Shoreland & CWH Assessment
  - Stakeholder Survey

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Water Quality Analysis

- General water chemistry (current & historical)
- Nutrient analysis
  - Lake trophic state (Eutrophication)
  - Limiting plant nutrient
- Supporting data for watershed modeling

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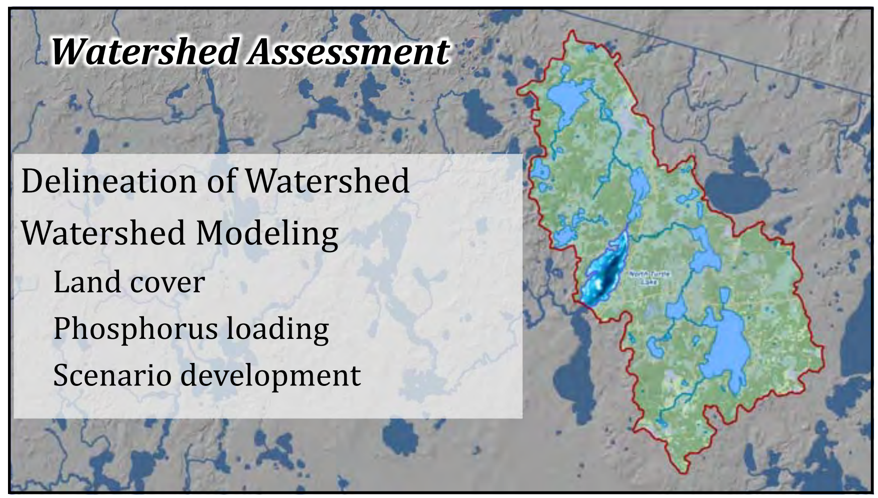


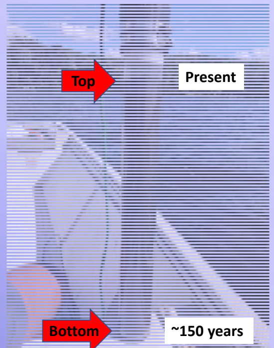
Watershed Assessment

Delineation of Watershed

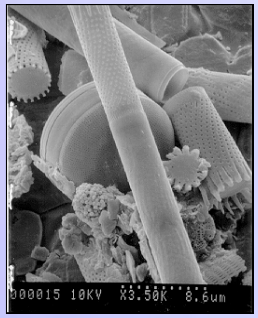
Watershed Modeling

- Land cover
- Phosphorus loading
- Scenario development

Paleocore Collection & Analysis



Sediment core

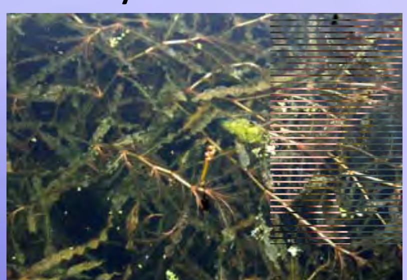

Diatoms

Aquatic Plant Surveys

- Concerned with both native and non-native plants
- Multiple surveys used in assessment
  - Early-Season AIS Survey (CLP, PYI, EWM)
  - Point-intercept survey
  - Emergent & floating-leaf community mapping

Non-native Aquatic Plants

Curly-leaf Pondweed



Verified 2020

Eurasian Watermilfoil

Verified 2003

Non-native Aquatic Plants

Pale Yellow Iris

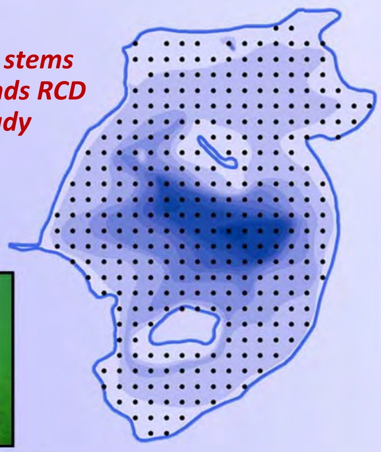
Verified 2017

Purple Loosestrife

Point-Intercept Survey

Collected EWM stems for Golden Sands RCD weevil study

Hatch Lake
37-meter Resolution
334 Total Points
WDNR Surveys: 2006



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Aquatic Plant Surveys

- Concerned with both native and non-native plants
- Multiple surveys used in assessment
  - Early-Season AIS Survey (CLP, PYI, EWM)
  - Point-intercept survey
  - Emergent & floating-leaf community mapping

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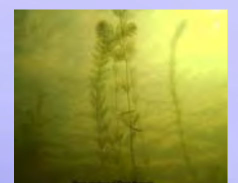
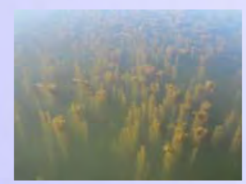
Professional AIS Mapping



Point-Based Mapping

- Single or Few Plants
- Clumps of Plants
- Small Plant Colony



Polygon-Based Mapping

- Highly Scattered
- Scattered
- Dominant
- Highly Dominant
- Surface Matting

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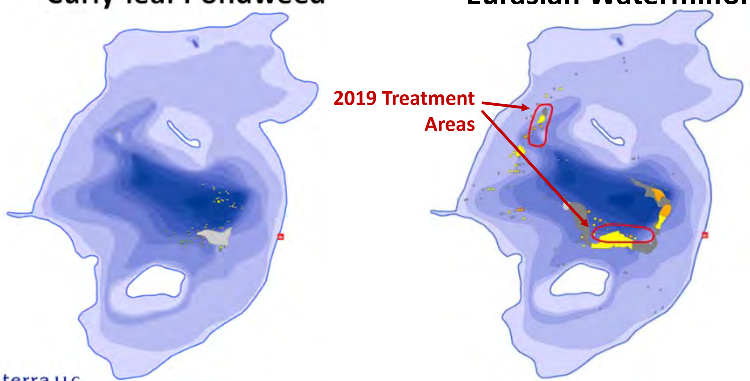
June 2020 Early-Season AIS Survey

Curly-leaf Pondweed **Eurasian Watermilfoil**



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Fisheries Data Integration

- No fish sampling completed
- Assemble data from WDNR, USGS, & USFWS
- Fish survey results summaries (if available)
- Use information in planning as applicable

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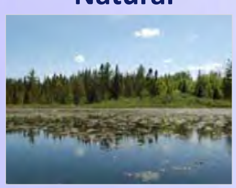
Shoreland Assessment

- Shoreland area is important for buffering runoff and provides valuable habitat for aquatic and terrestrial wildlife.
- EPA National Lakes Assessment results indicate shoreland development has greatest negative impact to health of our nation's lakes.
- It does not look at lake shoreline on a property-by-property basis.
- Assessment ranks shoreland area from shoreline back 35 feet

Urbanized

Range

Natural



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Stakeholder Survey

- Survey includes primarily riparian property owners
- Standard survey used as base
  - Planning committee potentially develops additional questions and options
  - Must not lead respondent to specific answer through a "loaded" question
- Survey must be approved by WDNR

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Planning Process

Planning Committee Meetings

Study Results (including a stakeholder survey)
Conclusions & Initial Recommendations
Management Goals
Management Actions
Timeframe
Facilitator(s)

Implementation Plan

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Thank You

Many of the graphics used in this presentation were supplied by:

Wisconsin Lakes Partnership
UW Extension
Wisconsin Dept. of Natural Resources

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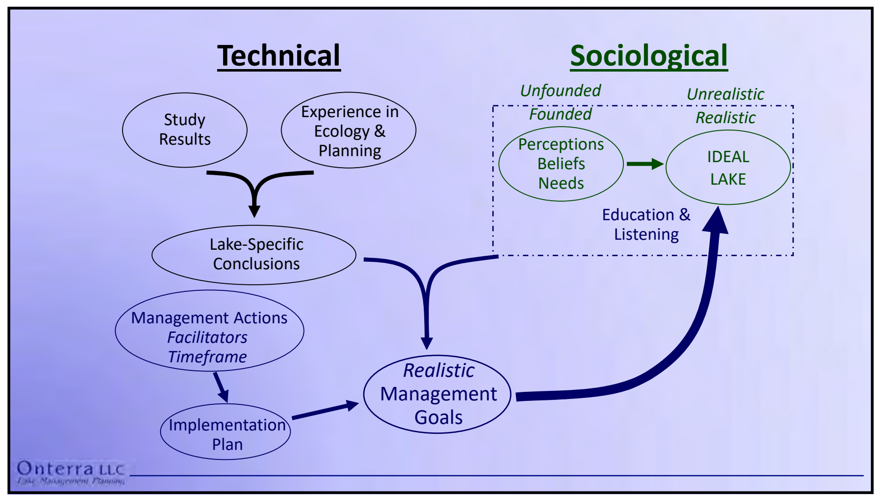

Planning Committee

- Role
  - Provide perspective as Hatch Lake stakeholder representatives
  - Gain understanding of Hatch Lake ecosystem and communicate with others
- Responsibilities
  - Stakeholder survey development (this summer)
  - Review draft result sections
  - Two planning meetings (2021)
  - Review/approve entire draft report
- Remember to record time spent on project activities (form provided)

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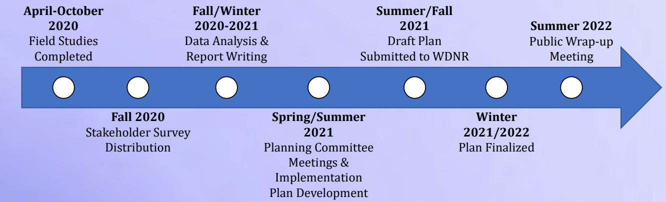
Project Timeline



- Next steps
  - Heather will be in touch soon regarding the stakeholder survey
  - Committee works with her to finalize survey – fall distribution
  - Field work completed through early 2021

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Presentation Outline

- Lake Management Planning Project Overview
- Meeting Objective
- Study Results
  - Water Quality
  - Paleoecology
  - Watershed
  - Shoreland Condition/Coarse Woody Habitat
  - Fishery
  - Aquatic Plants
- "Big Picture"
- Planning Meeting II

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Management Planning Project Overview

Collect and compile information about Hatch Lake

Includes both environmental & sociological

Historical & current information

Past management actions

Planning Meeting I
Report Sections

Create a realistic and implementable management plan

Challenges facing lake and HLA

Create goals that will address challenges

Develop actions that will meet goals

Assign timeframes & facilitators

Planning Meeting II
Implementation Plan

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Summary of Project Results

Water Quality

- Water quality is good and has been stable for the past 3 decades.
- Paleocore analysis may have picked up on some slight changes.

Watershed & Immediate Shoreline

- Watershed is small and in good condition.
- Changes in watershed and near shore zone would impact lake.

Fisheries

- Not much data available, but WDNR is scheduled to complete fishery survey starting fall 2019.

Aquatic Plant Community

- Aquatic plant community is healthy and of better than average quality

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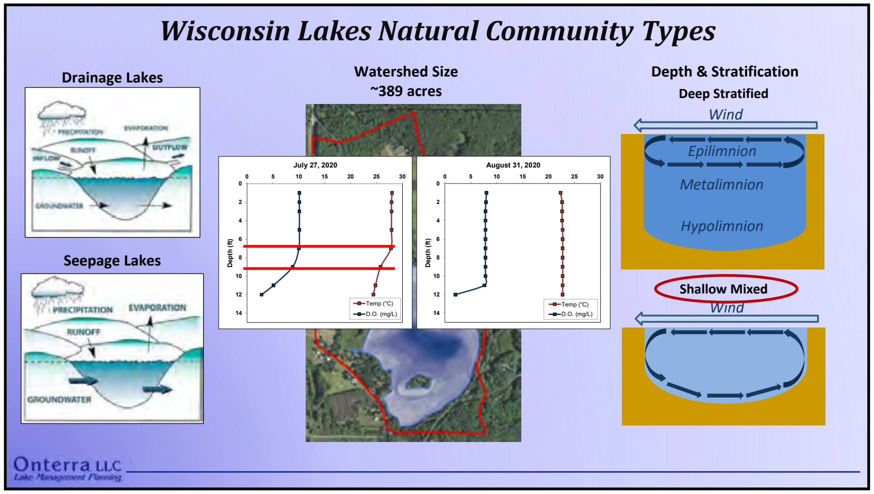
Wisconsin Lakes Natural Community Types
Drainage Lakes
Seepage Lakes
Watershed Size
~389 acres
July 27, 2020
August 31, 2020
Depth & Stratification
Deep Stratified
Wind
Epilimnion
Metalimnion
Hypolimnion
Shallow Mixed
Wind
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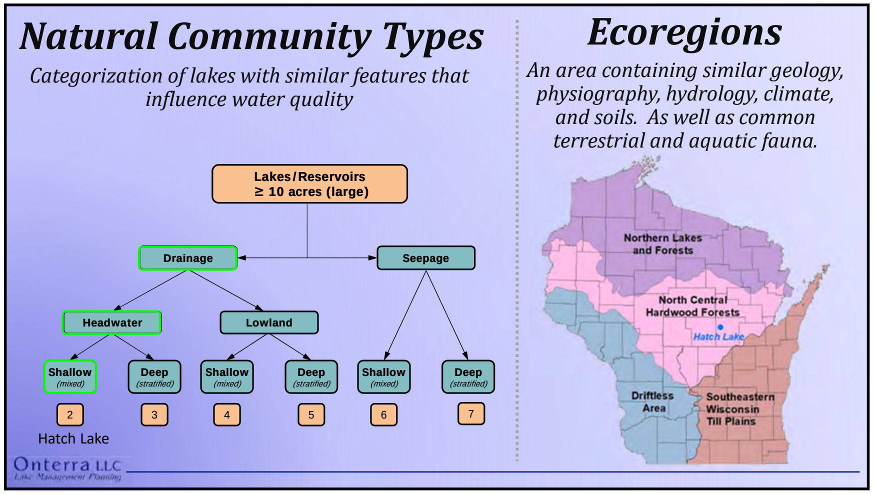
Natural Community Types
Categorization of lakes with similar features that influence water quality
Lakes/Reservoirs
≥ 10 acres (large)
Drainage
Seepage
Headwater
Lowland
Shallow (mixed)
Deep (stratified)
Shallow (mixed)
Deep (stratified)
Shallow (mixed)
Deep (stratified)
2
3
4
5
6
7
Hatch Lake
Ecoregions
An area containing similar geology, physiography, hydrology, climate, and soils. As well as common terrestrial and aquatic fauna.
Northern Lakes and Forests
North Central Hardwood Forests
Hatch Lake
Driftless Area
Southeastern Wisconsin Till Plains
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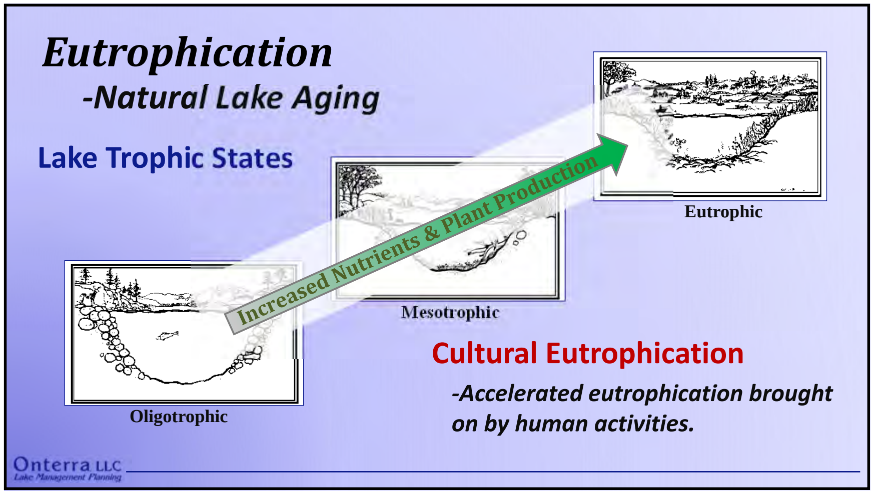
Eutrophication
-Natural Lake Aging
Lake Trophic States
Eutrophic
Mesotrophic
Oligotrophic
Increased Nutrients & Plant Production
Cultural Eutrophication
-Accelerated eutrophication brought on by human activities.
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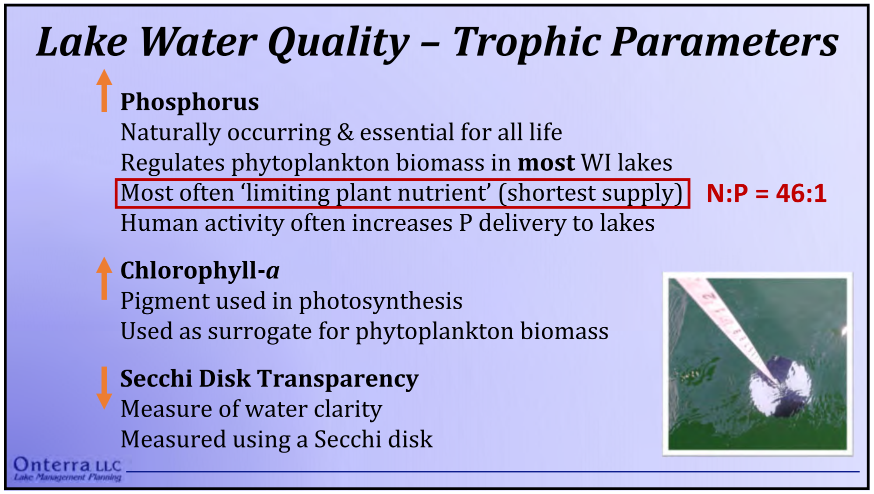
Lake Water Quality – Trophic Parameters
Phosphorus
Naturally occurring & essential for all life
Regulates phytoplankton biomass in most WI lakes
Most often 'limiting plant nutrient' (shortest supply)
N:P = 46:1
Human activity often increases P delivery to lakes
Chlorophyll-a
Pigment used in photosynthesis
Used as surrogate for phytoplankton biomass
Secchi Disk Transparency
Measure of water clarity
Measured using a Secchi disk
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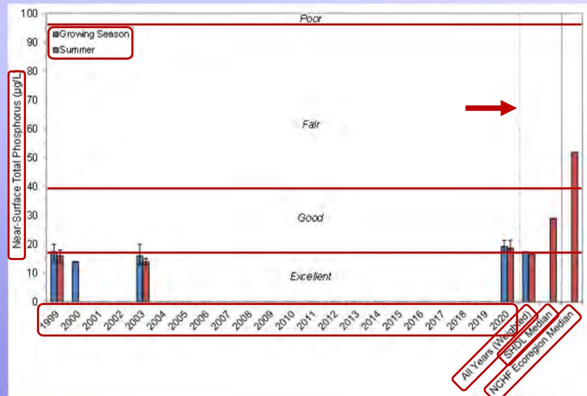
Hatch Lake Water Quality - Phosphorus



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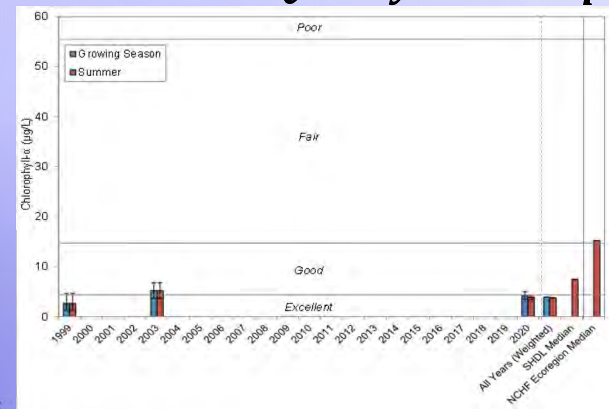
Hatch Lake Water Quality – Chlorophyll-a



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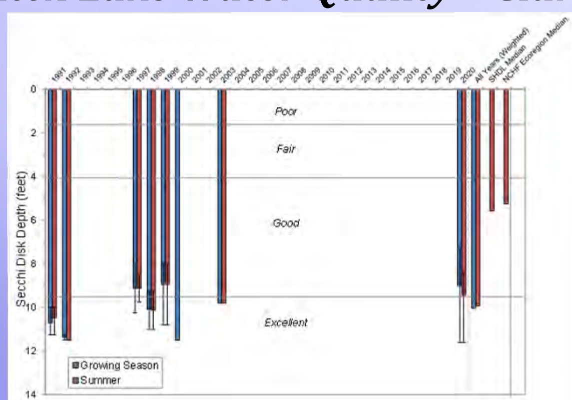
Hatch Lake Water Quality – Clarity



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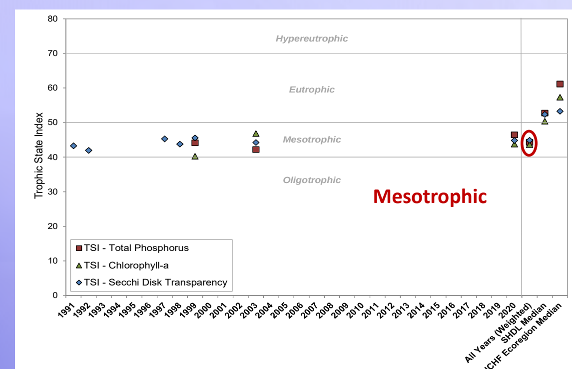
Hatch Lake Water Quality – Trophic State



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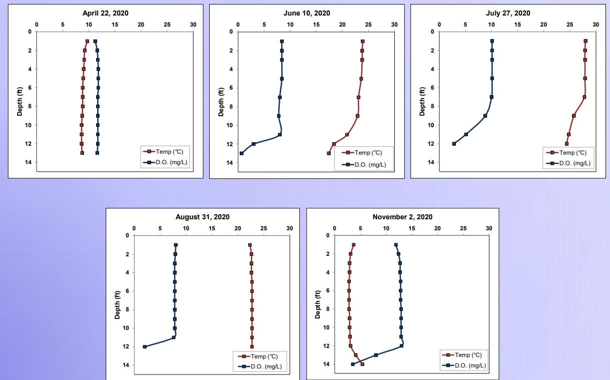
Dissolved Oxygen in Hatch Lake



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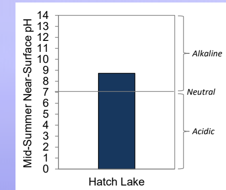
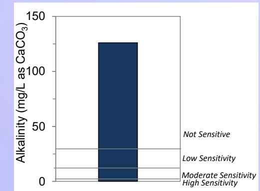
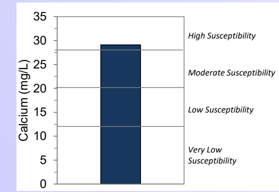
Additional Water Quality - Hatch Lake



Hatch is slightly alkaline



Hatch's high alkalinity means high buffering capacity against acid rain



High calcium levels mean that zebra mussels would establish well if introduced

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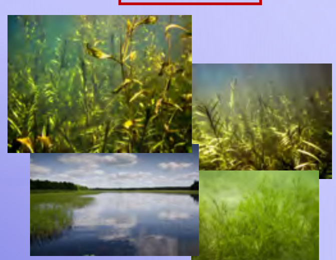
Shallow Lakes are Special

Clear State



Turbid State

Aquatic Plants are Incredibly Important

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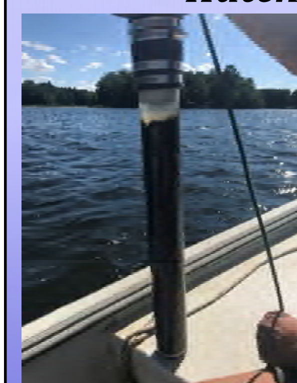
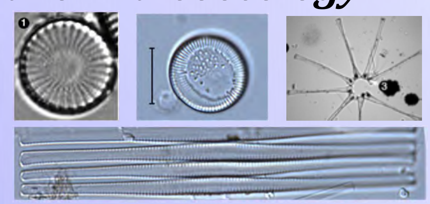
Hatch Lake - Paleoecology





Top-Bottom Sediment Core Results

- Plants likely occurred in basically the same areas, but are now more abundant and denser.
- Increase in vascular plants, and algae attached to them, have resulted in similar phosphorus concentrations between pre-European settlement and present.
- There is strong evidence of ecological degradation.

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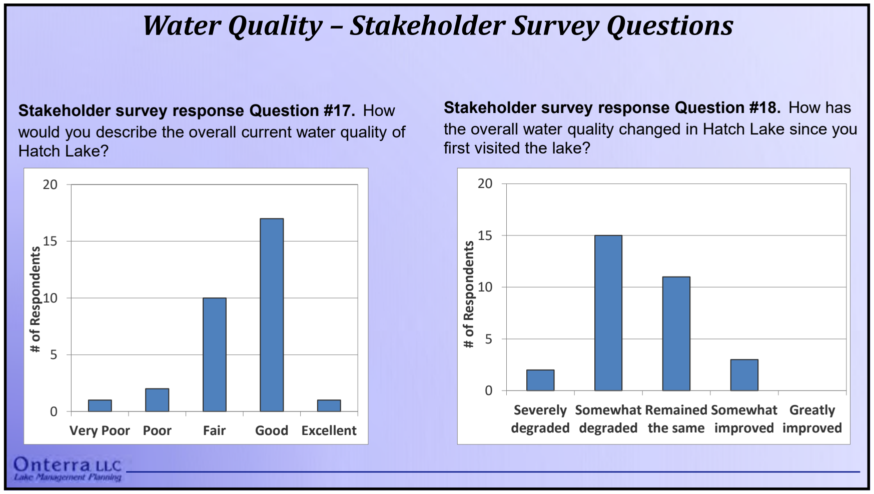
Water Quality – Stakeholder Survey Questions
Stakeholder survey response Question #17. How would you describe the overall current water quality of Hatch Lake?
# of Respondents
Very Poor
Poor
Fair
Good
Excellent
Stakeholder survey response Question #18. How has the overall water quality changed in Hatch Lake since you first visited the lake?
# of Respondents
Severely degraded
Somewhat degraded
Remained the same
Somewhat improved
Greatly improved
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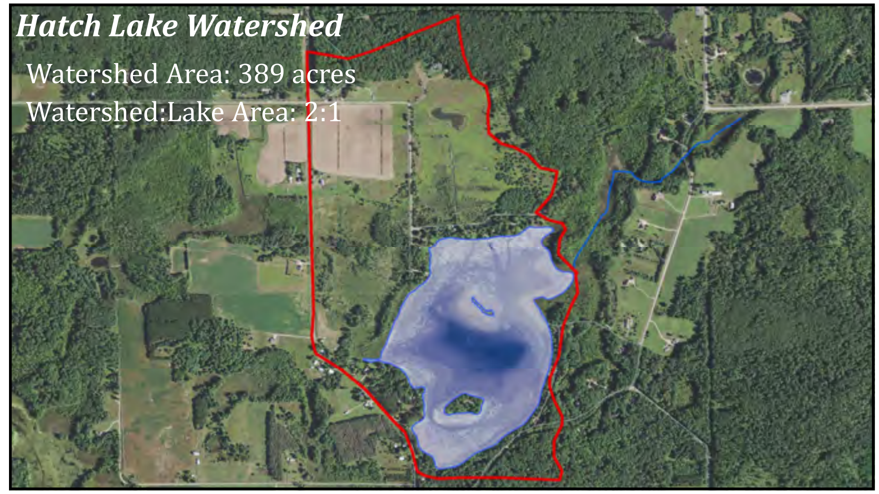
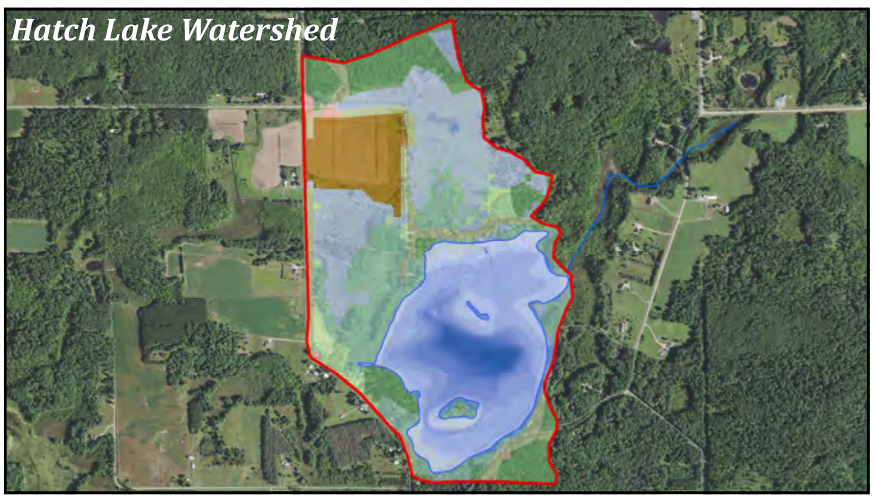
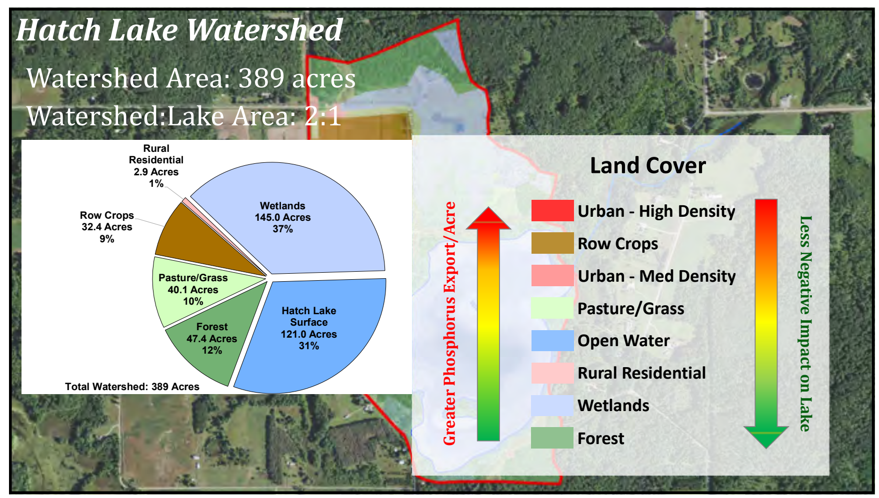
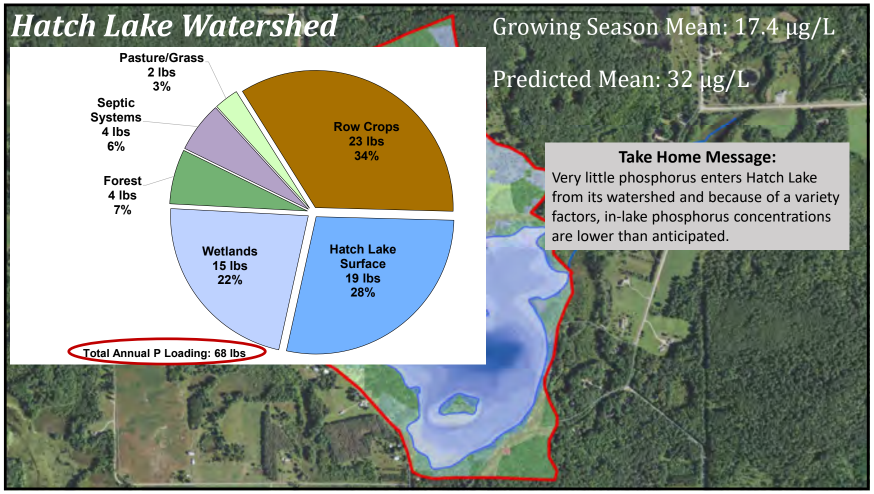

Hatch Lake Watershed
Watershed Area: 389 acres
Watershed:Lake Area: 2:1


Hatch Lake Watershed


Hatch Lake Watershed
Watershed Area: 389 acres
Watershed:Lake Area: 2:1
Rural Residential 2.9 Acres 1%
Row Crops 32.4 Acres 9%
Wetlands 145.0 Acres 37%
Pasture/Grass 40.1 Acres 10%
Forest 47.4 Acres 12%
Hatch Lake Surface 121.0 Acres 31%
Total Watershed: 389 Acres
Land Cover
Urban - High Density
Row Crops
Urban - Med Density
Pasture/Grass
Open Water
Rural Residential
Wetlands
Forest
Greater Phosphorus Export/Acre
Less Negative Impact on Lake


Hatch Lake Watershed
Growing Season Mean: 17.4 µg/L
Predicted Mean: 32 µg/L
Pasture/Grass 2 lbs 3%
Septic Systems 4 lbs 6%
Forest 4 lbs 7%
Row Crops 23 lbs 34%
Wetlands 15 lbs 22%
Hatch Lake Surface 19 lbs 28%
Total Annual P Loading: 68 lbs
Take Home Message:
Very little phosphorus enters Hatch Lake from its watershed and because of a variety factors, in-lake phosphorus concentrations are lower than anticipated.


Shoreland Assessment
• Shoreland area is important for buffering runoff and provides valuable habitat for aquatic and terrestrial wildlife.
• EPA National Lakes Assessment results indicate shoreland development has greatest negative impact to health of our nation's lakes.
• It does not look at lake shoreline on a property-by-property basis.
• Assessment ranks shoreland area from shoreline back 35 feet
Urbanized
Range
Natural
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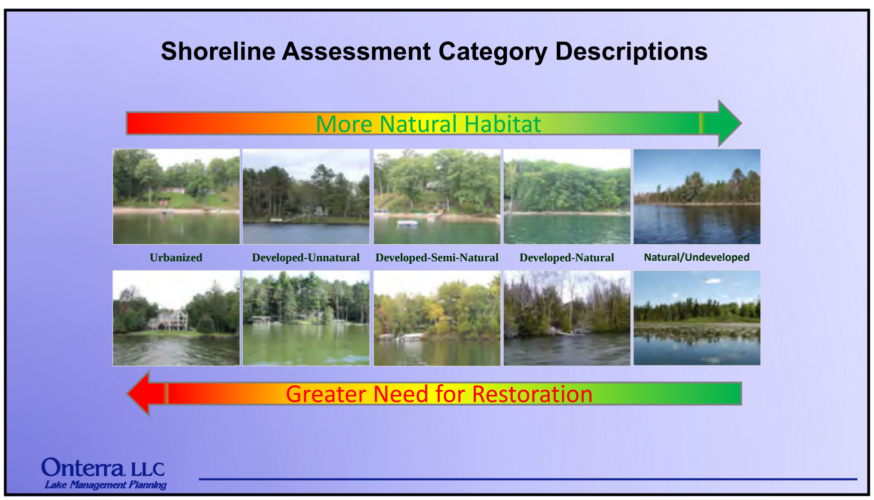
Shoreline Assessment Category Descriptions
More Natural Habitat
Urbanized
Developed-Unnatural
Developed-Semi-Natural
Developed-Natural
Natural/Undeveloped
Greater Need for Restoration
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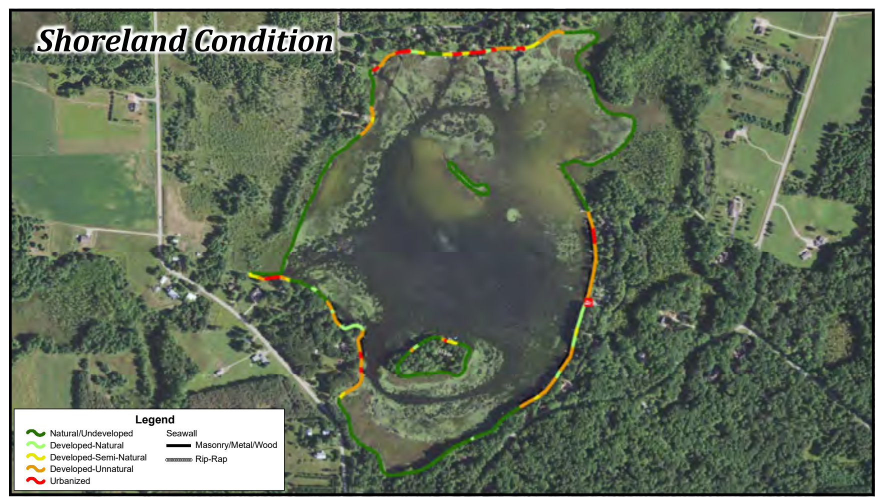
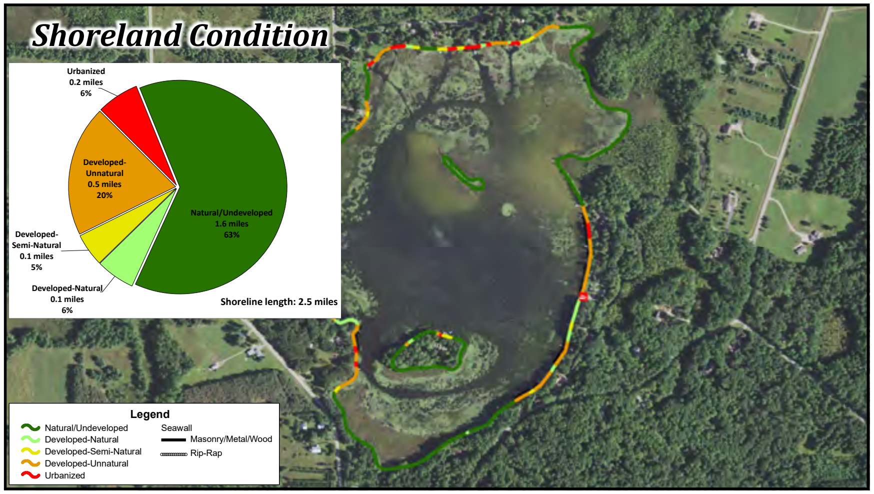
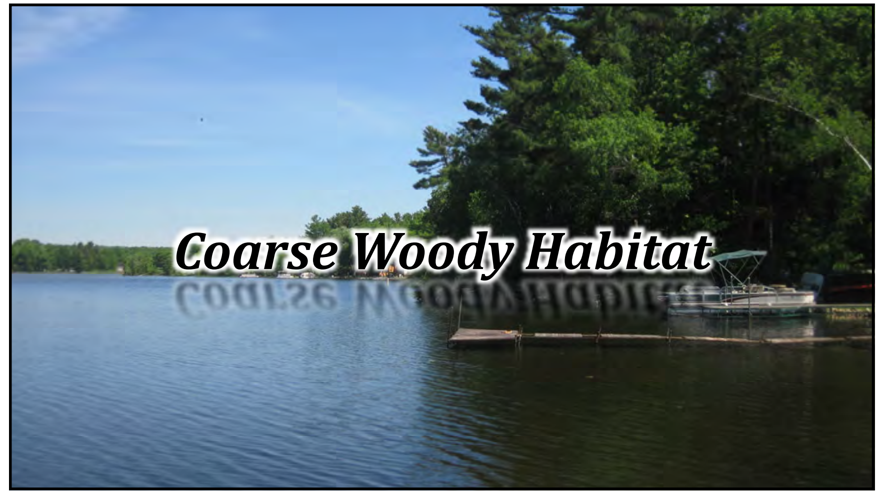
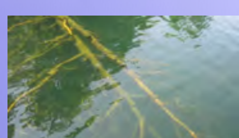

Shoreland Condition
Legend
Natural/Undeveloped
Developed-Natural
Developed-Semi-Natural
Developed-Unnatural
Urbanized
Seawall
Masonry/Metal/Wood
Rip-Rap

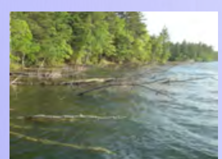




Coarse Woody Habitat

- Provides shoreland erosion control and prevents suspension of sediments.
- Preferred habitat for a variety of aquatic life.
  - Periphyton growth fed upon by insects.
  - Refuge, foraging and spawning habitat for fish.
  - Complexity of CWH important.
- Changing of logging and shoreland development practices = reduced CWH in Wisconsin lakes.
- Survey aimed at quantifying CWH in Town Lakes





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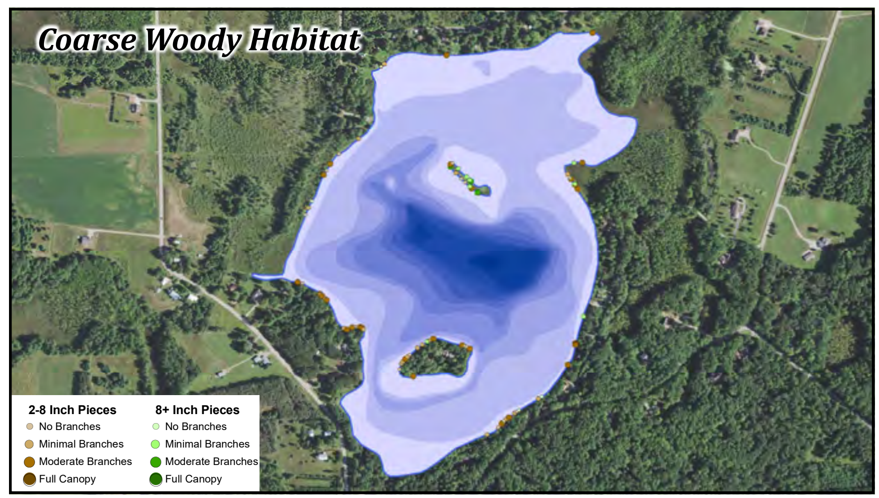
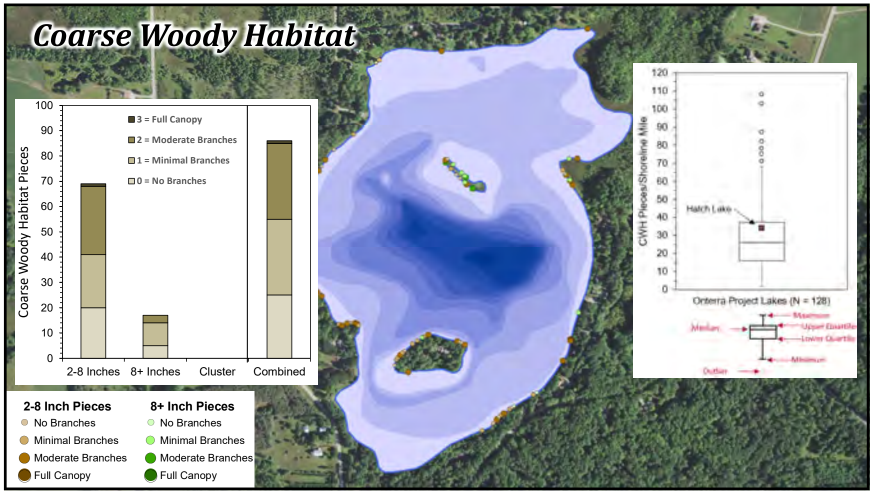
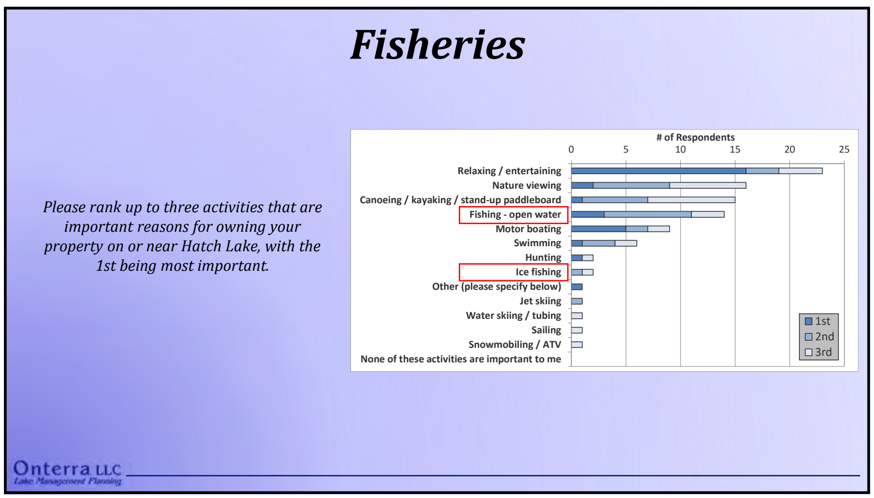



Coarse Woody Habitat
Coarse Woody Habitat Pieces
3 = Full Canopy
2 = Moderate Branches
1 = Minimal Branches
0 = No Branches
2-8 Inches
8+ Inches
Cluster
Combined
CWH Pieces/Shoreline Mile
Hatch Lake
Onterra Project Lakes (N = 128)
2-8 Inch Pieces
No Branches
Minimal Branches
Moderate Branches
Full Canopy
8+ Inch Pieces
No Branches
Minimal Branches
Moderate Branches
Full Canopy


Fisheries


Fisheries
Please rank up to three activities that are important reasons for owning your property on or near Hatch Lake, with the 1st being most important.
# of Respondents
Relaxing / entertaining
Nature viewing
Canoeing / kayaking / stand-up paddleboard
Fishing - open water
Motor boating
Swimming
Hunting
Ice fishing
Other (please specify below)
Jet skiing
Water skiing / tubing
Sailing
Snowmobiling / ATV
None of these activities are important to me
1st
2nd
3rd
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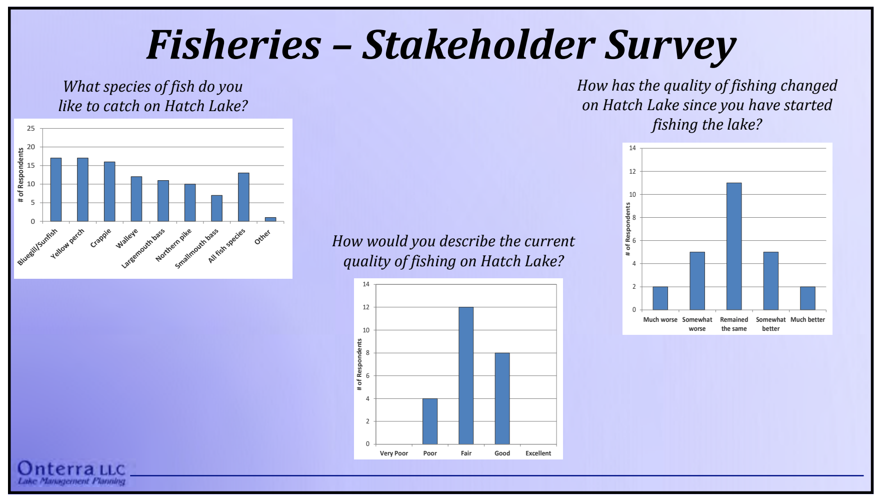
Fisheries – Stakeholder Survey
What species of fish do you like to catch on Hatch Lake?
How has the quality of fishing changed on Hatch Lake since you have started fishing the lake?
How would you describe the current quality of fishing on Hatch Lake?
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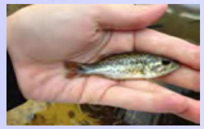
Fisheries

- History of fish kills led to installation of aeration system in 1989. Most recent WDNR comprehensive survey in 2014.
- Walleye stocked between 2005-2013, NOP stocked in 70's, LMB stocked in 1990 & 2017.
- WDNR Fisheries Biologist recommends focus on preserving habitat and water quality.
- LMB and walleye regulations in place to increase predator numbers and improve panfish growth and size structure.



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Aquatic Plant Surveys

- Assess both native and non-native populations
- Numerous surveys completed in 2020
  - Early-Season AIS Survey
  - Whole-Lake Point-Intercept Survey
  - Emergent/Floating-Leaf Community Mapping Survey

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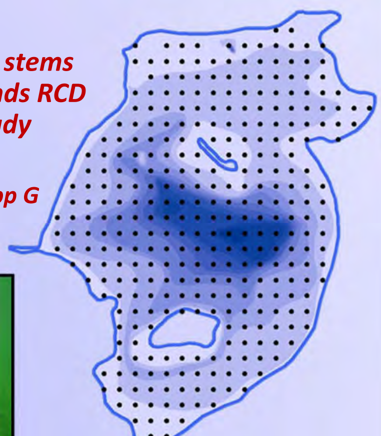
Lake Management Planning

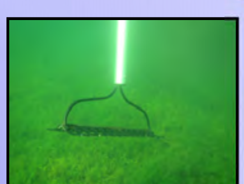
Point-Intercept Survey

Hatch Lake
37-meter Resolution
334 Total Points
WDNR Surveys: 2006

Collected EWM stems for Golden Sands RCD weevil study

- *Low Abundance*
- *Full Report in App G*





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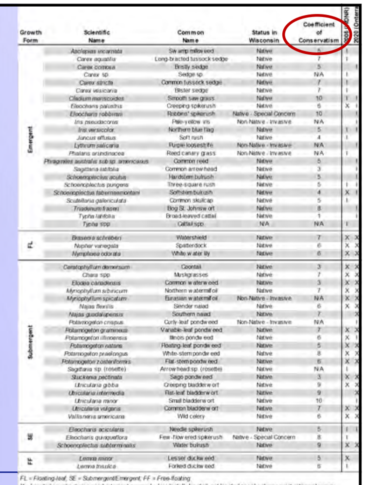
Aquatic Plant Species List

34 Native Species Total
21 Native Species on Rake
5 Non-Native Species
- Reed canary grass
- Purple loosestrife
- Pale-yellow iris
- Eurasian watermilfoil
- Curly-leaf pondweed

2 Special Concern Species
- Robbins' Spikerush
- Few-flowered Spikerush



Growth Form	Scientific Name	Common Name	Status in Wisconsin	Coefficient of Conservatism	2006 (WDNR)	2020 (Onterra)
Emergent	*Asclepias incarnata*	Swamp milkweed	Native	5	I	
	Carex aquatilis	Long-bracted tussock sedge	Native	7	I	
	Carex comosa	Bristly sedge	Native	5		I
	Carex sp.	Sedge sp.	Native	N/A	I	
	Carex stricta	Common tussock sedge	Native	7	I	
	Carex vesicaria	Blister sedge	Native	7	I	
	Cladium mariscoides	Smooth saw grass	Native	10	I	I
	Eleocharis palustris	Creeping spikerush	Native	6	X	I
	Eleocharis robbinsii	Robbins' spikerush	Native - Special Concern	10		I
	Iris pseudacorus	Pale-yellow iris	Non-Native - Invasive	N/A		I
	Iris versicolor	Northern blue flag	Native	5	I	I
	Juncus effusus	Soft rush	Native	4	I	
	Lythrum salicaria	Purple loosestrife	Non-Native - Invasive	N/A		I
	Phalaris arundinacea	Reed canary grass	Non-Native - Invasive	N/A	I	
	Phragmites australis subsp. *americanus*	Common reed	Native	5		I
	Sagittaria latifolia	Common arrowhead	Native	3		I
	Schoenoplectus acutus	Hardstem bulrush	Native	5		I
	Schoenoplectus pungens	Three-square rush	Native	5	I	I
	Schoenoplectus tabernaemontani	Softstem bulrush	Native	4	X	I
	Scutellaria galericulata	Common skullcap	Native	5	I	
	Triadenum fraseri	Bog St. John's-wort	Native	8		I
	Typha latifolia	Broad-leaved cattail	Native	1		I
	Typha spp.	Cattail spp.	N/A	N/A	I	
FL	*Brasenia schreberi*	Watershield	Native	7	X	X
	Nuphar variegata	Spatterdock	Native	6	X	X
	Nymphaea odorata	White water lily	Native	6	X	X
Submergent	*Ceratophyllum demersum*	Coontail	Native	3	X	X
	Chara spp.	Muskgrasses	Native	7	X	X
	Elodea canadensis	Common waterweed	Native	3	X	X
	Myriophyllum sibiricum	Northern watermilfoil	Native	7	X	X
	Myriophyllum spicatum	Eurasian watermilfoil	Non-Native - Invasive	N/A	X	X
	Najas flexilis	Slender naiad	Native	6	X	X
	Najas guadalupensis	Southern naiad	Native	7		X
	Potamogeton crispus	Curly-leaf pondweed	Non-Native - Invasive	N/A		I
	Potamogeton gramineus	Variable-leaf pondweed	Native	7	X	X
	Potamogeton illinoensis	Illinois pondweed	Native	6	X	I
	Potamogeton natans	Floating-leaf pondweed	Native	5	X	X
	Potamogeton praelongus	White-stem pondweed	Native	8	X	X
	Potamogeton zosteriformis	Flat-stem pondweed	Native	6	X	X
	Sagittaria sp. (rosette)	Arrowhead sp. (rosette)	Native	N/A	I	
	Stuckenia pectinata	Sago pondweed	Native	3	X	X
	Utricularia gibba	Creeping bladderwort	Native	9	X	X
	Utricularia intermedia	Flat-leaf bladderwort	Native	9		X
	Utricularia minor	Small bladderwort	Native	10		I
	Utricularia vulgaris	Common bladderwort	Native	7	X	X
	Vallisneria americana	Wild celery	Native	6	X	X
SE	*Eleocharis acicularis*	Needle spikerush	Native	5	I	I
	Eleocharis quinqueflora	Few-flowered spikerush	Native - Special Concern	8	I	
	Schoenoplectus subterminalis	Water bulrush	Native	9	X	X
FF	*Lemna minor*	Lesser duckweed	Native	5	X	
	Lemna trisulca	Forked duckweed	Native	6	I	

FL = Floating-leaf; SE = Submergent/Emergent; FF = Free-floating
X = Located on rake during point-intercept survey; I = Incidentally located, not located on rake during point-intercept survey

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Vegetation Analysis Matrices

Floristic Quality Analysis

Evaluates the closeness of an area's flora to undisturbed conditions.

$$I = \bar{C} \times \sqrt{N}$$

I Floristic Quality Index

$\bar{C}$ Average Species Conservatism
1 – 10, higher number requires less disturbed condition

N Number of Native Species
Only species encountered on the rake are used (no incidentals)

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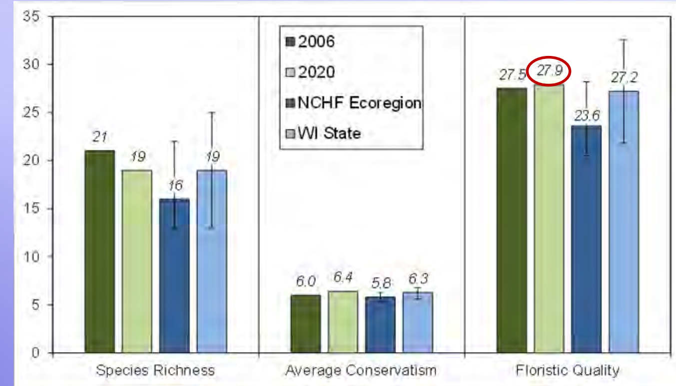
Floristic Quality Analysis



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Vegetation Analysis Matrices

Species Diversity

Species diversity utilizes species richness and also takes into account evenness or the variation in abundance of the individual species within the community.

A community of 10 species with the population evenly divided among those species is more diverse than a community of 10 species with 50% of the population in one or two species.

A more diverse community can withstand environmental fluctuations better than a less diversity community.

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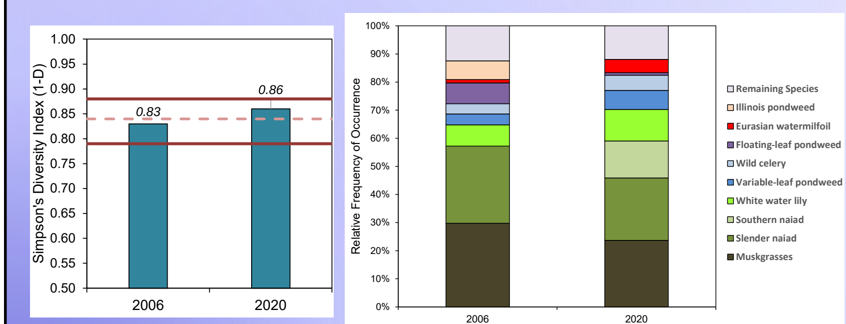
Simpson's Diversity Index



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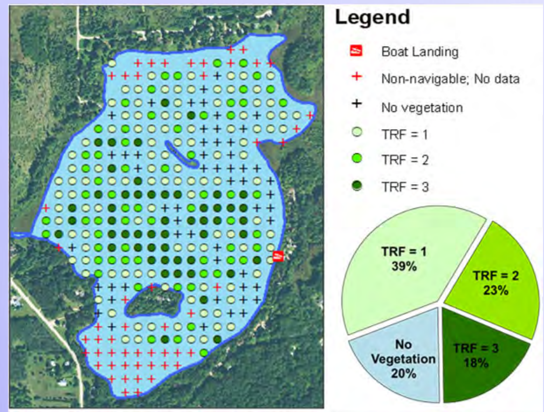
Total Rake Fullness

Rake-fullness = 1

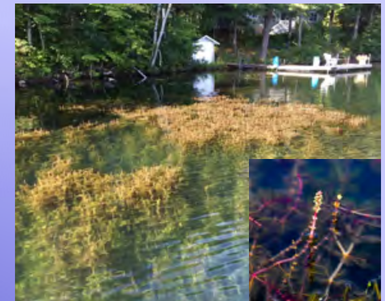
Rake-fullness = 2

Rake-fullness = 3



Non-Native Aquatic Plants

Eurasian Watermilfoil



Curly-leaf Pondweed

Professional AIS Mapping

Point-Based Mapping

- Single plants to colonies or areas less than 40-feet in diameter
- Abundance descriptions:
  - Single or Few Plants
  - Clumps of Plants
  - Small Plant Colony

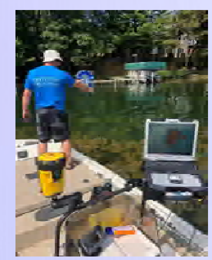
Professional AIS Mapping

Polygon-Based Mapping

- Colonies or areas over 40-feet diameter
- Boundary at target plant extent or morphological feature (depth contour, shoreline)
- Density ratings:

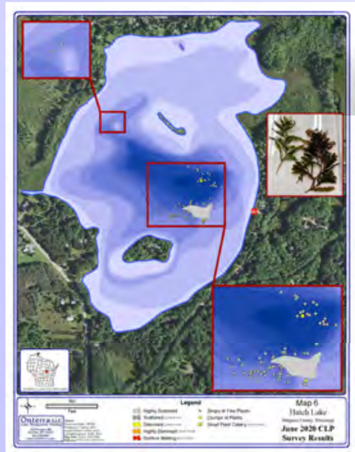
May not represent true colonies or "beds"
- Highly Scattered
- Scattered

Increase in Ecological Impact
- Dominant
- Highly Dominant
- Surface Matting

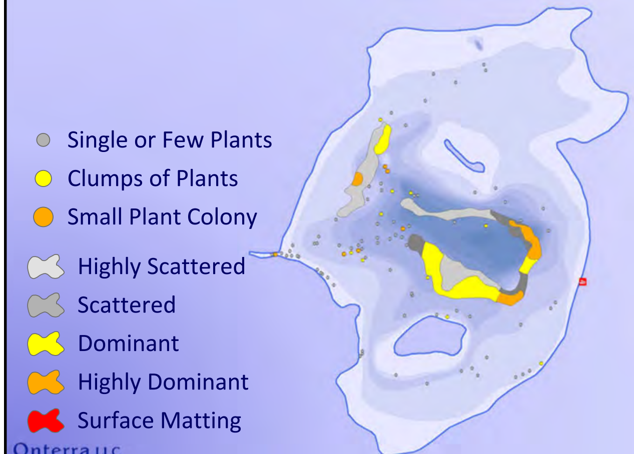


June 2020 CLP Survey Results

Not previously documented in lake, likely present for some time



2020 Late-Season Mapping Results



Hatch Lake
Waupaca County
106 Acres

EWM Management History

Verified in Hatch Lake in 2003, likely there prior

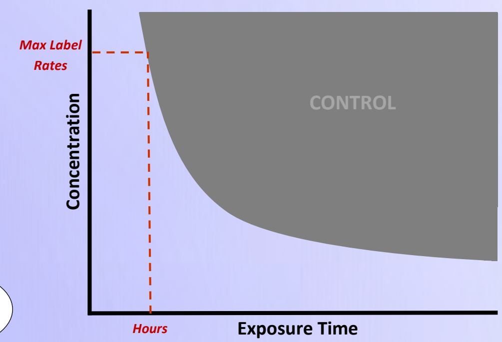
Herbicide Spot Treatments:
2013 - 3.5 acres – 2,4-D
2016 – 1.7 acres – 2,4-D & endothall
2019 – 1.75 & 1.25 acres – Aquastrike™ (diquat + endothall)

Seasonal EWM control expected

Ecological Definitions of Herbicide Treatment

Spot Treatment:

Herbicide applied at a scale where dissipation will not result in significant lake wide concentrations; impacts are anticipated to be localized to in/around application area.



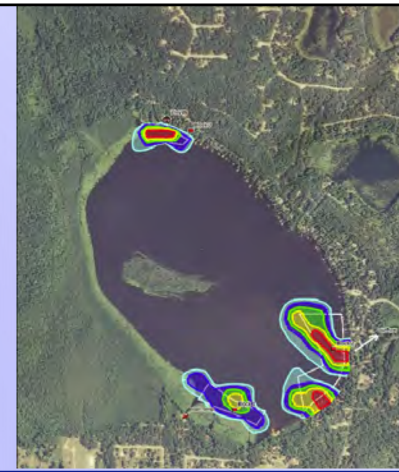
High Concentration ▶ Short Exposure Time

2015 Treatment on Loon Lake

- **Diquat (2 gallons per surface acre of application area)**
- ~24 acres of 305 acre lake (7.8%)
- Tracer Dye (Rhodamine WT) Survey

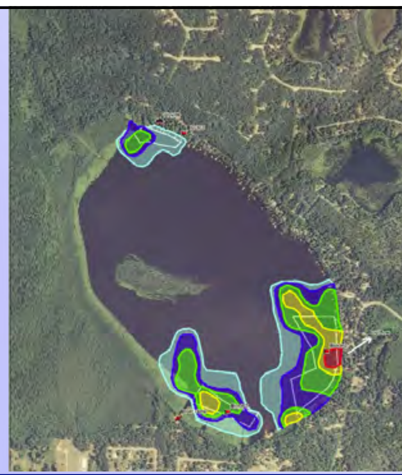
1 HAT

75-100%
50-75%
25-50%
10-25%
5-10%



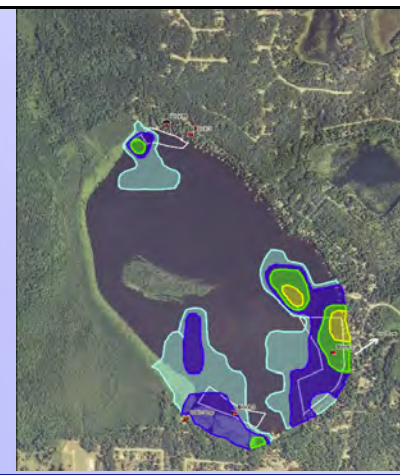
2.5 HAT

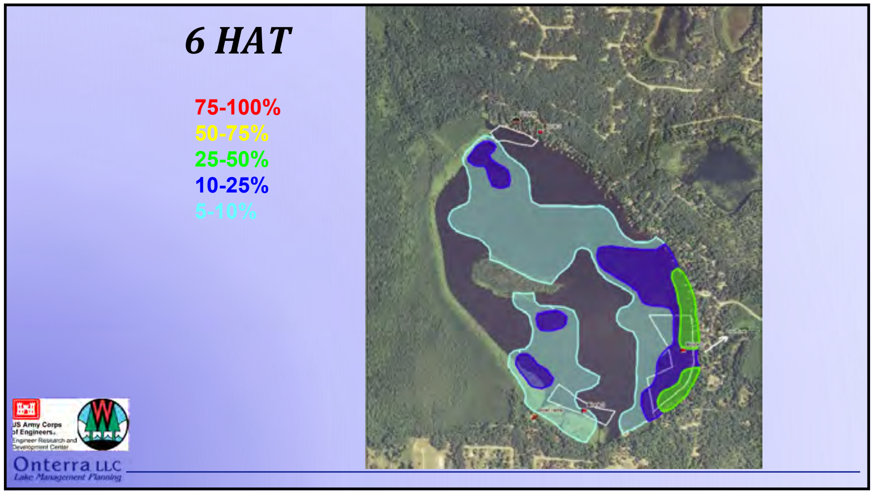
75-100%
50-75%
25-50%
10-25%
5-10%

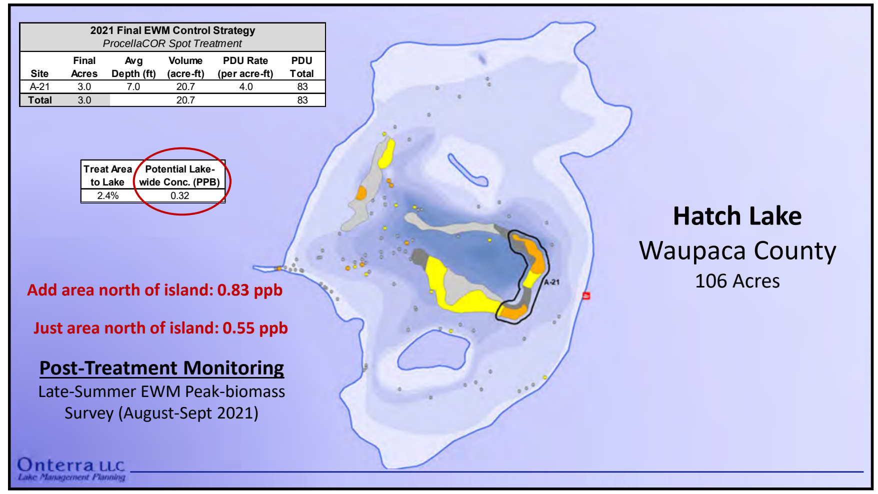


4 HAT

75-100%
50-75%
25-50%
10-25%
5-10%







Conclusions

Water Quality

- Water quality is good and as expected.
- Limited data prevents long-term analysis.

Watershed & Immediate Shoreline

- Limited development on shorelands and high quality landcover lead to very good water quality and habitat value.

Aquatic Plant Community

- Aquatic plant community is of high quality.
- While there are abundant plants, they do aid in water quality.
- Exotic species are at low levels and controllable.

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Planning Meeting II

Primary Objective: Create implementation plan framework

Steps to Achieve Objective:

1. Discuss challenges facing lake and lake group
2. Convert challenges to management goals
3. Create management actions to meet management goals
4. Determine timeframes and facilitators to carry out actions

Assignment for Planning Meeting II

1. Create list of challenges facing lake and lake group (keep to yourself)
2. Review stakeholder survey results (Tim! - Handout)
3. Send potential report section edits and questions to Tim

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Thank You

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Lake Management Planning

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Management Planning Project Overview

Collect and compile information about Hatch Lake
- *Includes both environmental & sociological*
- *Historical & current information*
- *Past management actions*

Planning Meeting I
Report Sections

Create a realistic and implementable management plan
- *Challenges facing lake and HLA*
- *Create goals that will address challenges*
- *Develop actions that will meet goals*
- *Assign timeframes & facilitators*

Planning Meeting II
Implementation Plan

Onterra LLC Lake Management Planning

Conclusions

Water Quality
- Water quality is good and as expected.
- Limited data prevents long-term analysis.

Watershed & Immediate Shoreline
- Limited development on shorelands and high quality landcover lead to very good water quality and habitat value.

Aquatic Plant Community
- Aquatic plant community is of high quality.
- While there are abundant plants, they do aid in water quality.
- Exotic species are at low levels and controllable.

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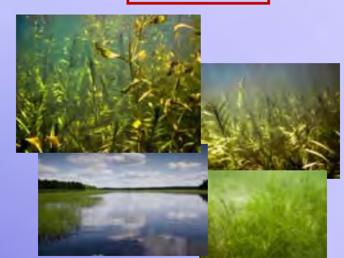
Shallow Lakes are Special

Clear State



Aquatic Plants are Incredibly Important

Turbid State

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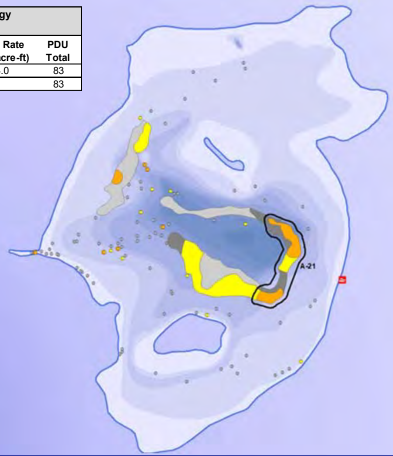
2021 Final EWM Control Strategy *ProcellaCOR Spot Treatment*					
Site	Final Acres	Avg Depth (ft)	Volume (acre-ft)	PDU Rate (per acre-ft)	PDU Total
A-21	3.0	7.0	20.7	4.0	83
Total	3.0		20.7		83

Treat Area to Lake	Potential Lake-wide Conc. (PPB)
2.4%	0.32



Hatch Lake
Waupaca County
106 Acres

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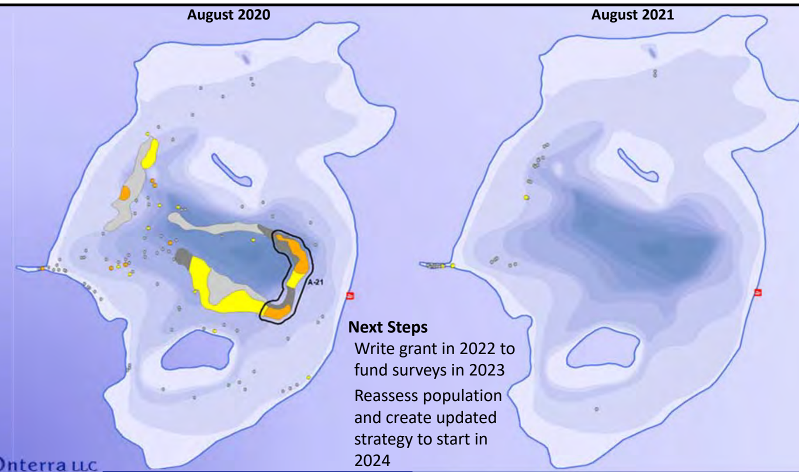

Next Steps
Write grant in 2022 to fund surveys in 2023
Reassess population and create updated strategy to start in 2024

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Thank You

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B

APPENDIX B

Stakeholder Survey Response Charts and Comments

Hatch Lake - Anonymous Stakeholder Survey

Surveys Distributed: 43
Surveys Returned: 31
Response Rate: 72%

Hatch Lake Property

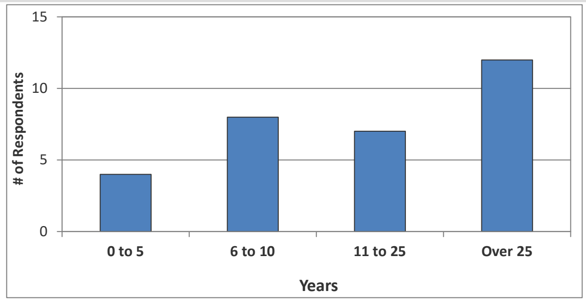
1. Is your property on the lake or off the lake?

Answer Options	Response Percent	Response Count
On the lake	100.0%	31
Off the lake	0.0%	0
	answered question	**31**
	skipped question	**0**

2. How many years have you owned your property on or near Hatch Lake?

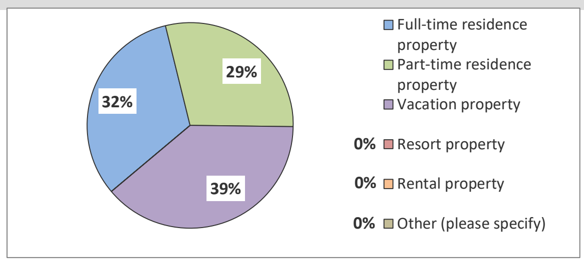
Answer Options	Response Count
	31
answered question	**31**
skipped question	**0**

Category (# of years)	Responses	% Response
0 to 5	**4**	**13%**
6 to 10	**8**	**26%**
11 to 25	**7**	**23%**
Over 25	**12**	**39%**



3. How is your property on or near Hatch Lake used?

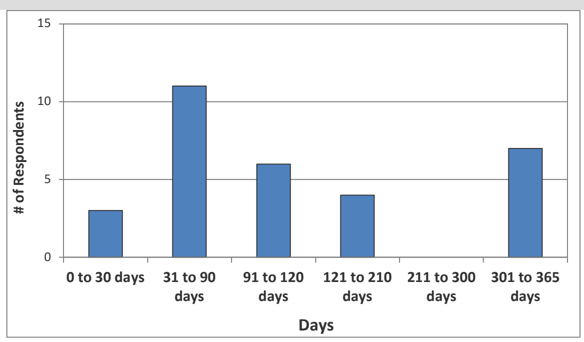
Answer Options	Response Percent	Response Count
Full-time residence property	32.3%	10
Part-time residence property	29.0%	9
Vacation property	38.7%	12
Resort property	0.0%	0
Rental property	0.0%	0
Other (please specify)	0.0%	0
	answered question	**31**
	skipped question	**0**



4. Considering the past three years, how many days each year is your property used by you or others?

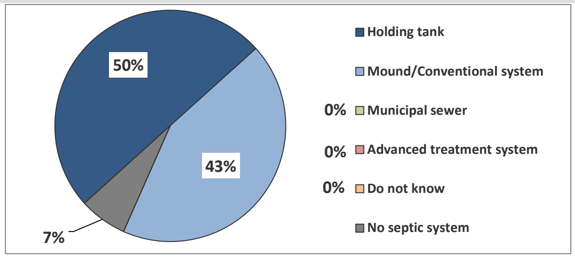
	Response Count
answered question	**31**
skipped question	**0**

Category (# of days)	Responses	
0 to 30 days	**3**	**10%**
31 to 90 days	**11**	**35%**
91 to 120 days	**6**	**19%**
121 to 210 days	**4**	**13%**
211 to 300 days	**0**	**0%**
301 to 365 days	**7**	**23%**



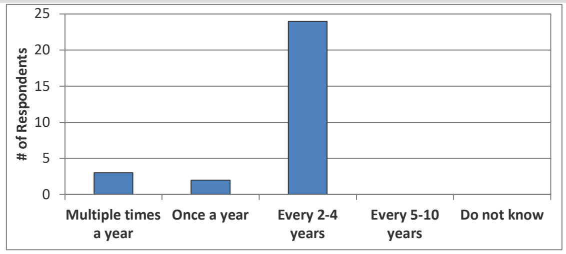
5. What type of septic system does your property have?

Answer Options	Response Percent	Response Count
Holding tank	50.0%	15
Mound/Conventional system	43.3%	13
Municipal sewer	0.0%	0
Advanced treatment system	0.0%	0
Do not know	0.0%	0
No septic system	6.7%	2
	answered question	30
	skipped question	1



6. How often is the septic system on your property pumped?

Answer Options	Response Percent	Response Count
Multiple times a year	10.3%	3
Once a year	6.9%	2
Every 2-4 years	82.8%	24
Every 5-10 years	0.0%	0
Do not know	0.0%	0
	answered question	29
	skipped question	2

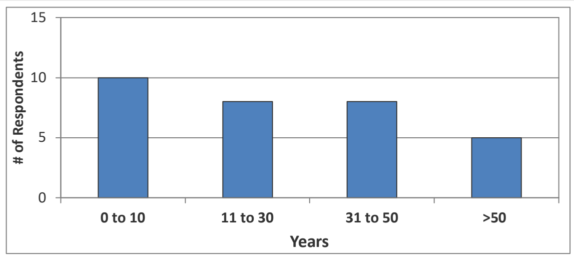


<u>Recreational Activity on Hatch Lake</u>

7. How many years ago did you first visit Hatch Lake?

Answer Options	Response Count
answered question	31
skipped question	0

Category (# of years)	**Responses**	**% Response**
0 to 10	**10**	**32%**
11 to 30	**8**	**26%**
31 to 50	**8**	**26%**
>50	**5**	**16%**

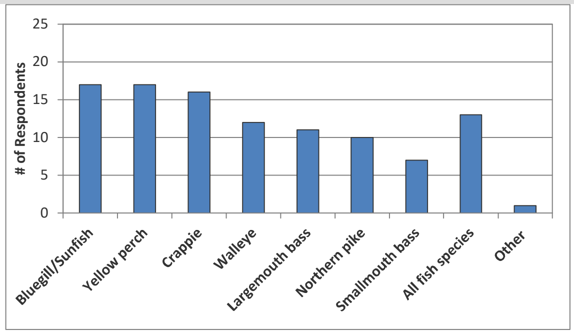


8. Have you personally fished on Hatch Lake in the past three years?

Answer Options	Response Percent	Response Count
Yes	80.7%	25
No	19.4%	6
	answered question	31
	skipped question	0

9. What species of fish do you like to catch on Hatch Lake?

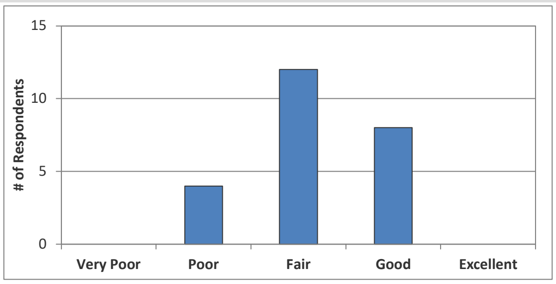
Answer Options	Response Percent	Response Count
Bluegill/Sunfish	68.0%	17
Yellow perch	68.0%	17
Crappie	64.0%	16
Walleye	48.0%	12
Largemouth bass	44.0%	11
Northern pike	40.0%	10
Smallmouth bass	28.0%	7
All fish species	52.0%	13
Other	4.0%	1
	answered question	25
	skipped question	6



Number	Other (please specify)
1	bullhead

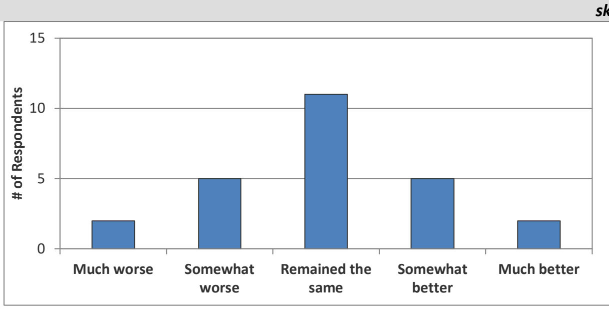
10. How would you describe the current quality of fishing on Hatch Lake?

Answer Options	Very Poor	Poor	Fair	Good	Excellent	Response Count
	0	4	12	8	0	
					answered question	24
					skipped question	7



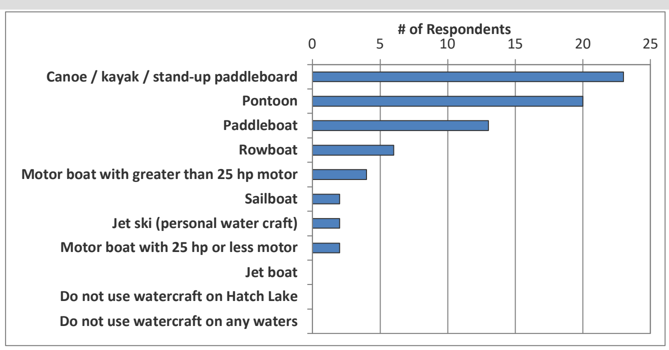
11. How has the quality of fishing changed on Hatch Lake since you have started fishing the lake?

Answer Options	Much worse	Somewhat worse	Remained the same	Somewhat better	Much better	Response Count
	2	5	11	5	2	
					answered question	25
					skipped question	6



12. What types of watercraft do you currently use on Hatch Lake?

Answer Options	Response Percent	Response Count
Canoe / kayak / stand-up paddleboard	74.2%	23
Pontoon	64.5%	20
Paddleboat	41.9%	13
Rowboat	19.4%	6
Motor boat with greater than 25 hp motor	12.9%	4
Sailboat	6.5%	2
Jet ski (personal water craft)	6.5%	2
Motor boat with 25 hp or less motor	6.5%	2
Jet boat	0.0%	0
Do not use watercraft on Hatch Lake	0.0%	0
Do not use watercraft on any waters	0.0%	0
	answered question	31
	skipped question	0



13. Do you use your watercraft on waters other than Hatch Lake?

Answer Options	Response Percent	Response Count
Yes	3.2%	1
No	96.8%	30
	answered question	**31**
	skipped question	**0**

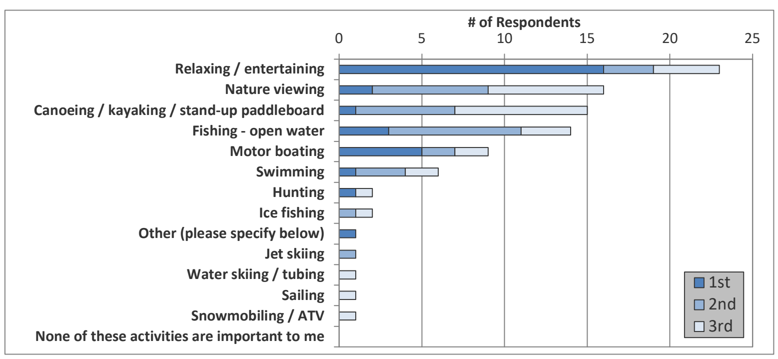
14. What is your typical cleaning routine after using your watercraft on waters other than Hatch Lake?

Answer Options	Response Percent	Response Count
Remove aquatic hitch-hikers (ex. - plant material, clams, mussels)	100.0%	1
Drain bilge	100.0%	1
Rinse boat	100.0%	1
Power wash boat	0.0%	0
Apply bleach	0.0%	0
Air dry boat for 5 or more days	0.0%	0
Do not clean boat	0.0%	0
Other (please specify)	0.0%	0
	answered question	**1**
	skipped question	**30**

15. Please rank up to three activities that are important reasons for owning your property on or near Hatch Lake, with the 1st being most important.

Answer Options	1st	2nd	3rd	Weighted Average	Response Count
Relaxing / entertaining	16	3	4	1.48	23
Nature viewing	2	7	7	2.31	16
Canoeing / kayaking / stand-up paddleboard	1	6	8	2.47	15
Fishing - open water	3	8	3	2	14
Motor boating	5	2	2	1.67	9
Swimming	1	3	2	2.17	6
Hunting	1	0	1	2	2
Ice fishing	0	1	1	2.5	2
Other (please specify below)	1	0	0	1	1
Jet skiing	0	1	0	2	1
Water skiing / tubing	0	0	1	3	1
Sailing	0	0	1	3	1
Snowmobiling / ATV	0	0	1	3	1
None of these activities are important to me	0	0	0	0	0
				answered question	**31**
				skipped question	**0**

Number	"Other" responses
1	pontooning
2	Lake free of mud spots

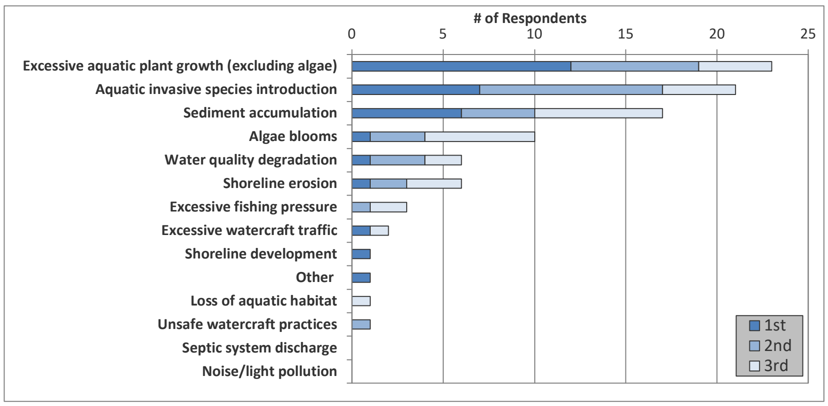


Hatch Lake Current and Historic Condition, Health and Management

16. From the list below, please rank your top three concerns regarding Hatch Lake, with the 1st being your top concern.

Answer Options	1st	2nd	3rd	Response Count
Excessive aquatic plant growth (excluding algae)	12	7	4	23
Aquatic invasive species introduction	7	10	4	21
Sediment accumulation	6	4	7	17
Algae blooms	1	3	6	10
Water quality degradation	1	3	2	6
Shoreline erosion	1	2	3	6
Excessive fishing pressure	0	1	2	3
Excessive watercraft traffic	1	0	1	2
Shoreline development	1	0	0	1
Other	1	0	0	1
Loss of aquatic habitat	0	0	1	1
Unsafe watercraft practices	0	1	0	1
Septic system discharge	0	0	0	0
Noise/light pollution	0	0	0	0
			answered question	31
			skipped question	0

Number	"Other" responses
1	The bottom has detached and has floated to the top in multiple areas.

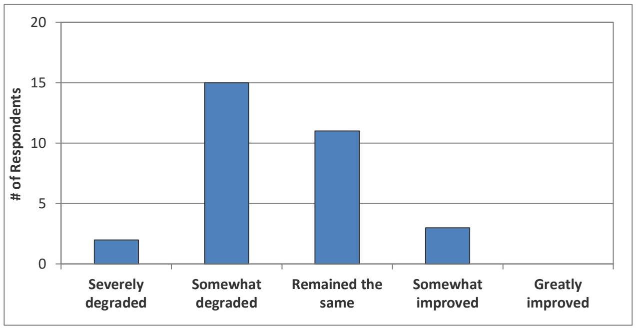


17. How would you describe the overall current water quality of Hatch Lake?

Answer Options	Very Poor	Poor	Fair	Good	Excellent	Response Count
	1	2	10	17	1	
					answered question	31
					skipped question	0

18. How has the overall water quality changed in Hatch Lake since you first visited the lake?

Answer Options	Severely degraded	Somewhat degraded	Remained the same	Somewhat improved	Greatly improved	Response Count
	2	15	11	3	0	
					answered question	**31**
					skipped question	**0**



19. Which of the following would you say is the single most important aspect when considering water quality?

Answer Options	Response Percent	Response Count
Aquatic plant growth (not including algae blooms)	54.8%	17
Water clarity (clearness of water)	41.9%	13
Smell	3.2%	1
Water color	0.0%	0
Algae blooms	0.0%	0
Water level	0.0%	0
Fish kills	0.0%	0
Other (please specify)	0.0%	0
	answered question	**31**
	skipped question	**0**

20. Before reading the statement above, had you ever heard of aquatic invasive species?

Answer Options	Response Percent	Response Count
Yes	100.0%	31
No	0.0%	0
	answered question	**31**
	skipped question	**0**

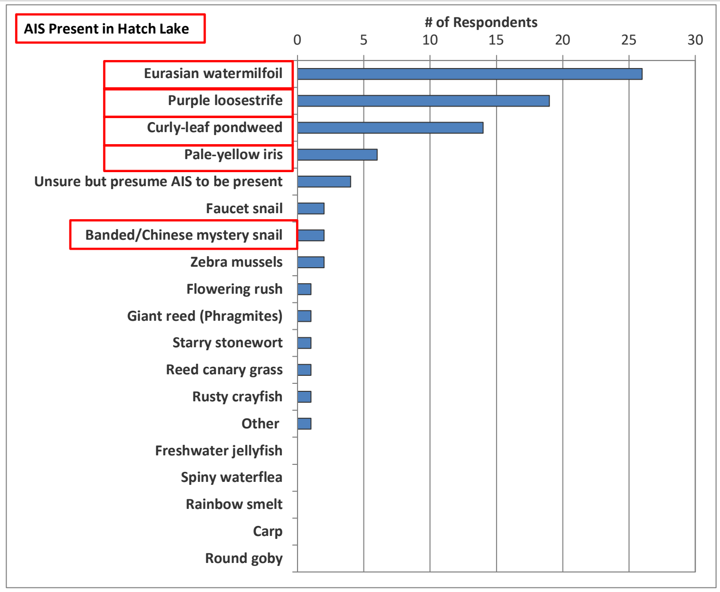
21. Do you believe aquatic invasive species are present within Hatch Lake?

Answer Options	Response Percent	Response Count
Yes	80.7%	25
I think so but am not certain	16.1%	5
No	3.2%	1
	answered question	**31**
	skipped question	**0**

22. Which aquatic invasive species do you believe are present in or immediately around Hatch Lake?

Answer Options	Response Percent	Response Count
Eurasian watermilfoil	86.7%	26
Purple loosestrife	63.3%	19
Curly-leaf pondweed	46.7%	14
Pale-yellow iris	20.0%	6
Unsure but presume AIS to be present	13.3%	4
Faucet snail	6.7%	2
Banded/Chinese mystery snail	6.7%	2
Zebra mussels	6.7%	2
Flowering rush	3.3%	1
Giant reed (Phragmites)	3.3%	1
Starry stonewort	3.3%	1
Reed canary grass	3.3%	1
Rusty crayfish	3.3%	1
Other	3.3%	1
Freshwater jellyfish	0.0%	0
Spiny waterflea	0.0%	0
Rainbow smelt	0.0%	0
Carp	0.0%	0
Round goby	0.0%	0
	answered question	**30**
	skipped question	**1**

Number	"Other" responses
1	buckthorn, barberry, honeysuckle, white suckers, bullhead



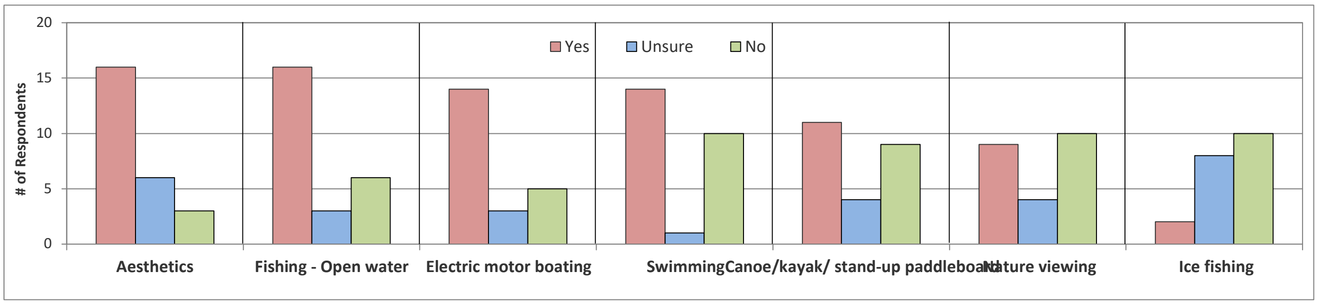
23. Do you believe you are able to identify Eurasian watermilfoil in Hatch Lake?

Answer Options	Response Percent	Response Count
Yes	58.1%	18
I think so but am not certain	25.8%	8
No	16.1%	5
	answered question	**31**
	skipped question	**0**

24. Has the Eurasian watermilfoil population ever had a negative impact on your enjoyment of Hatch Lake?

Answer Options	Yes	Unsure	No	Response Count
Aesthetics	16	6	3	25
Fishing - Open water	16	3	6	25
Electric motor boating	14	3	5	22
Swimming	14	1	10	25
Canoe/kayak/ stand-up paddleboard	11	4	9	24
Nature viewing	9	4	10	23
Ice fishing	2	8	10	20
Other				1
			answered question	26
			skipped question	5

Number	"Other" responses
1	fishing - great fish habitat, but tough difficult to fish (fish get tangled in weeds)



25. Before the present year, aquatic herbicides have been used to manage Eurasian watermilfoil on Hatch Lake. Professional monitoring of the aquatic plant community has also occurred during this time. Prior to reading this information, did you know that aquatic herbicides were being applied in Hatch Lake to manage Eurasian watermilfoil?

Answer Options	Response Percent	Response Count
Yes	80.7%	25
I think so but am not certain	12.9%	4
No	6.5%	2
	answered question	31
	skipped question	0

26. What is your level of support or opposition for the past use of aquatic herbicides to treat Eurasian watermilfoil in Hatch Lake in previous years?

Answer Options	Completely support	Moderately support	Neutral	Moderately oppose	Completely oppose	Response Count
	18	5	5	3	0	
					answered question	31
					skipped question	0

27. What is your level of support or opposition for the future use of aquatic herbicides to treat Eurasian watermilfoil in Hatch Lake?

Answer Options	Completely support	Moderately support	Neutral	Moderately oppose	Completely oppose	Response Count
	16	7	5	2	1	
					answered question	31
					skipped question	0

28. If you selected "Moderately oppose" or "Completely oppose" for Question #27, what is the reason or reasons you oppose the future use of aquatic herbicides to target Eurasian watermilfoil in Hatch Lake?

Answer Options	Response Percent	Response Count
Potential cost of treatment is too high.	100.0%	3
Potential impacts to native aquatic plant species	66.7%	2
Potential impacts to human health	66.7%	2
Ineffectiveness of herbicide strategy	66.7%	2
Potential impacts to native (non-plant) species such as fish, insects, etc.	33.3%	1
Future impacts are unknown	33.3%	1
Another reason (please specify)	0.0%	0
	answered question	3
	skipped question	28

Hatch Lake Association (HLA)

29. Before receiving this mailing, had you ever heard of the HLA?

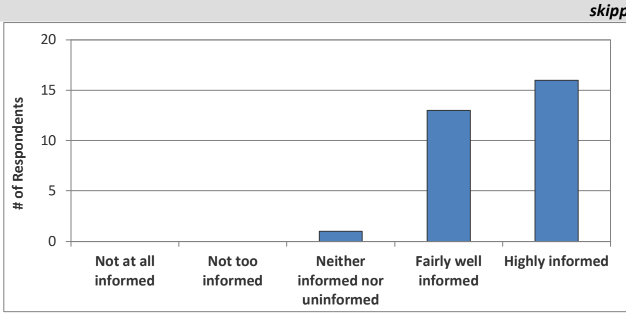
Answer Options	Response Percent	Response Count
Yes	96.8%	30
No	3.2%	1
	answered question	**31**
	skipped question	**0**

30. What is your membership status with the HLA?

Answer Options	Response Percent	Response Count
Current member	100.0%	30
Former member	0.0%	0
Never been a member	0.0%	0
	answered question	**30**
	skipped question	**1**

31. How informed has (or had) the HLA kept you regarding issues with Hatch Lake and its management?

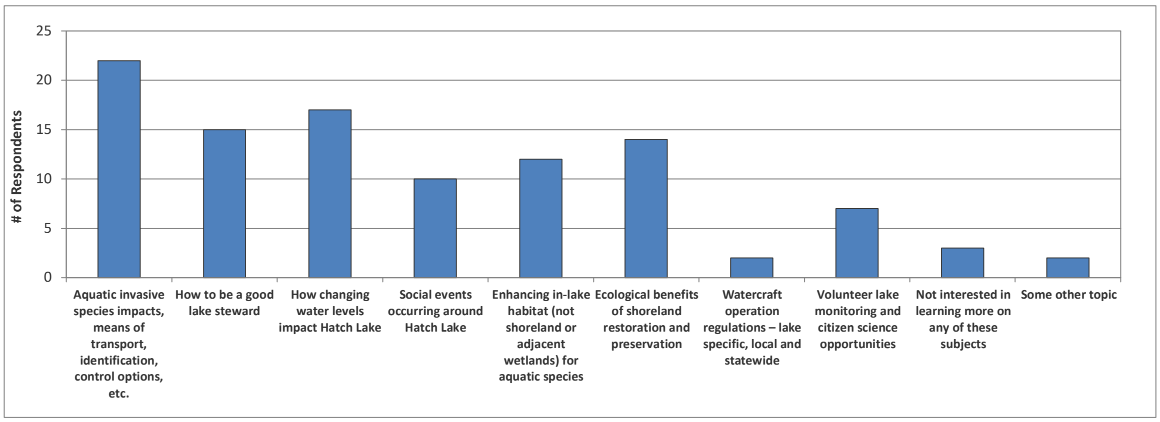
Answer Options	Not at all informed	Not too informed	Neither informed nor uninformed	Fairly well informed	Highly informed	Response Count
	0	0	1	13	16	
					answered question	**30**
					skipped question	**1**



32. Stakeholder education is an important component of every lake management planning effort. Which of these subjects would you like to learn more about?

Answer Options	Response Percent	Response Count
Aquatic invasive species impacts, means of transport, identification, control options, etc.	73.3%	22
How to be a good lake steward	50.0%	15
How changing water levels impact Hatch Lake	56.7%	17
Social events occurring around Hatch Lake	33.3%	10
Enhancing in-lake habitat (not shoreland or adjacent wetlands) for aquatic species	40.0%	12
Ecological benefits of shoreland restoration and preservation	46.7%	14
Watercraft operation regulations – lake specific, local and statewide	6.7%	2
Volunteer lake monitoring and citizen science opportunities	23.3%	7
Not interested in learning more on any of these subjects	10.0%	3
Some other topic	6.7%	2
	answered question	30
	skipped question	1

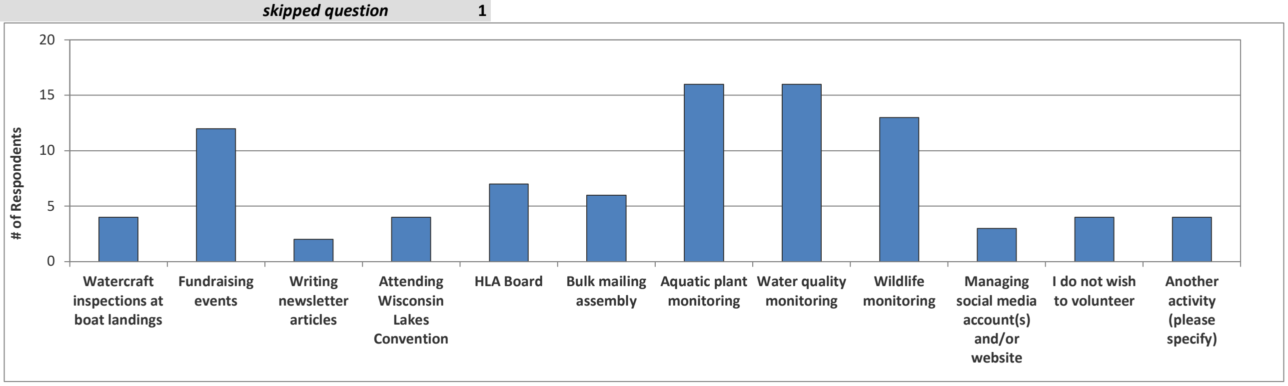
Number	Other (please specify)
1	how get rid of some of the muck, not all of but in spots where needed
2	Organic natural options to do invasive weed control in safe environment



33. The effective management of Hatch Lake will require the cooperative efforts of numerous volunteers. Please select the activities you would be willing to participate in if the HLA requires additional assistance.

Answer Options	Response Percent	Response Count
Watercraft inspections at boat landings	13.3%	4
Fundraising events	40.0%	12
Writing newsletter articles	6.7%	2
Attending Wisconsin Lakes Convention	13.3%	4
HLA Board	23.3%	7
Bulk mailing assembly	20.0%	6
Aquatic plant monitoring	53.3%	16
Water quality monitoring	53.3%	16
Wildlife monitoring	43.3%	13
Managing social media account(s) and/or webs	10.0%	3
I do not wish to volunteer	13.3%	4
Another activity (please specify)	13.3%	4
	answered question	30
	skipped question	1

Number	Other (please specify)
1	muck removal
2	lake clean ups
3	volunteering at fundraisers
4	Already on fishing comity



34. Please feel free to provide written comments concerning Hatch Lake, its current and/or historic condition, and its management.

Answer Options	Response Count
answered question	12
skipped question	19

Number	Response Text
1	To the management team, keep up the good work all you guys and ladies.
2	would like to see weeds controlled more before they take over the whole lake.
3	Thanks for organizing this survey to continue to improve Hatch Lake!
4	Warner level and water quality
5	For a small organization we are able to get a lot done. It's important to know your neighbors and this organization helps facilitate that.
6	Love Hatch Lake and especially the HLA. It brings us all together to get to know each other, share our concerns, and work on solutions as a group. The summer picnic is GREAT!
7	Since the start of Hatch Lake Association we had a strong commitment to better the lake. When we first moved here the lake would have terrible fish kills, not much wild life lived here at all. In fact we had geese as our first logo because that was the only thing that lived on the lake and there were not very many, now they are a pest and our logo now has change to a loon. There are now swans, snow geese, multiple different ducks that stop in the spring, eagles all year round. So if look back 40 years the lake has come a long ways from where it was. Boat use be able to race all over the place anytime they wanted, now we have a no wake from 5 at night until 10 in the morning for some quite time. So all these short times on this lake don't let them bull shit ya to much, the lake has come a long ways over the years.
8	Is additional aeration systems on the bad sides of the lake an option? before certain areas become unpassable.
9	- things are moving in right direction overall - it would be nice to be as concerned about terrestrial invasive species as much as aquatic ones - not sure why fish are stocked that are already present and abundant within the lake (especially if already stunted) - i.e. LM bass - it would be nice to stock walleye to improve panfish stunting and to introduce another species - tighter panfish limits (10/day) would be great if they help panfish stunting - it would be nice to have a firmer base in more areas - i.e. remove some of the sediment/muck - it seems like the lake has always had great vegetation, thought lily pads are increasing - winter aeration is a wonderful thing for the lake
10	In 1989 I was able to drive my boat to all of Hatch Lakes shores to fish and explore. Lilly pad growth and the resulting sediment from those and other weeds have since made 50 percent of our shoreline un navigable. And now the milfoil is trying to take the middle of the lake. Many years ago I suggested an aggressive attack on Lilly pads. It never happened and the result has devastated those parts of the lake. I feel we must take an aggressive approach to the milfoil or loose dozens of acres to another nasty weed. Hatch Lake has a good variety of plantlife to fill the needs of its Aquatic critters. We can get by very well with no EM and much less lillypad acreage.
11	The water along our shoreline is full of muck that smells awful and has an overgrowth of lily pads and milfoil. There used to be fish all around us. Now there are very few. It smells awful
12	Not in favor of chemicals used in our lake to treat invasive species. We should continue lake cleanup days and we need more volunteers.

C

APPENDIX C

Water Quality Data

	Secchi (feet)				Chlorophyll-*a* (µg/L)				Total Phosphorus (µg/L)			
	Growing Season		Summer		Growing Season		Summer		Growing Season		Summer	
Year	**Count**	**Mean**	**Count**	**Mean**	**Count**	**Mean**	**Count**	**Mean**	**Count**	**Mean**	**Count**	**Mean**
1991	12	10.7	8	10.5								
1992	2	11.4	1	11.5								
1993	0		0									
1994	0		0									
1995	0		0									
1996	0		0									
1997	4	9.1	2	9.1								
1998	6	10.1	4	10.1								
1999	3	8.9	3	8.9	3	2.7	3	2.7	3	17.3	2.0	16.0
2000	1	11.5	0		0		0		1	14.0	0.0	
2001	0		0		0		0		0		0.0	
2002	0		0		0		0		0		0.0	
2003	1	9.8	1	9.8	2	5.2	2	5.2	3	16.0	2.0	14.0
2004	0		0		0		0		0		0.0	
2005	0		0		0		0		0		0.0	
2006	0		0		0		0		0		0.0	
2007	0		0		0		0		0		0.0	
2008	0		0		0		0		0		0.0	
2009	0		0		0		0		0		0.0	
2010	0		0		0		0		0		0.0	
2011	0		0		0		0		0		0.0	
2012	0		0		0		0		0		0.0	
2013	0		0		0		0		0		0.0	
2014	0		0		0		0		0		0.0	
2015	0		0		0		0		0		0.0	
2016	0		0		0		0		0		0.0	
2017	0		0		0		0		0		0.0	
2018	0		0		0		0		0		0.0	
2019	0		0		0		0		0		0.0	
2020	5	9.0	4	9.4	4	4.1	3	3.8	4	19.3	3.0	18.8
All Years (Weighted)		10.0		9.9		3.9		3.7		17.4		16.6
SHDL Median				5.6				7.5				29.0
NCHF Ecoregion Median				5.3				15.2				52.0

D

APPENDIX D

Point-Intercept Aquatic Macrophyte Survey Data

	Scientific Name	Common Name	LFOO (%)	
			2006	2020
Dicots	*Nymphaea odorata*	White water lily	16.7	17.0
	Myriophyllum spicatum	Eurasian watermilfoil	2.6	7.1
	Utricularia gibba	Creeping bladderwort	7.9	2.5
	Myriophyllum sibiricum	Northern watermilfoil	7.9	1.1
	Nuphar variegata	Spatterdock	3.3	2.1
	Utricularia vulgaris	Common bladderwort	2.3	0.7
	Ceratophyllum demersum	Coontail	0.3	1.1
	Brasenia schreberi	Watershield	0.3	1.1
	Utricularia intermedia	Flat-leaf bladderwort	0.0	1.1
Non-dicots	*Najas flexilis & N. guadalupensis*	Slender & Southern naiads	61.0	46.8
	Chara spp.	Muskgrasses	65.9	35.8
	Najas flexilis	Slender naiad	61.0	33.7
	Potamogeton gramineus & P. illinoensis	Variable-leaf & Illinois pondweeds	22.0	10.3
	Najas guadalupensis	Southern naiad	0.0	19.9
	Potamogeton gramineus	Variable-leaf pondweed	8.5	10.3
	Vallisneria americana	Wild celery	8.2	8.2
	Potamogeton natans	Floating-leaf pondweed	16.4	1.4
	Potamogeton illinoensis	Illinois pondweed	14.8	0.0
	Schoenoplectus subterminalis	Water bulrush	2.0	4.6
	Elodea canadensis	Common waterweed	3.6	2.8
	Potamogeton praelongus	White-stem pondweed	3.6	2.1
	Stuckenia pectinata	Sago pondweed	1.0	1.1
	Potamogeton zosteriformis	Flat-stem pondweed	1.6	0.4
	Schoenoplectus tabernaemontani	Softstem bulrush	0.7	0.0
	Lemna minor	Lesser duckweed	0.3	0.0
	Fissidens spp. & Fontinalis spp.	Aquatic Moss	0.3	0.0
	Eleocharis palustris	Creeping spikerush	0.3	0.0

E

APPENDIX E

2014 Spring Electrofishing (SEII) Summary Report Hatch Lake, Waupaca County

Hatch Lake (WBIC 282800)

Waupaca County

Introduction and Survey Objectives

In 2014, the Department of Natural Resources conducted a one night boomshocking survey of Hatch Lake in order to provide insight and direction for the future fisheries management of this water body. Primary sampling objectives of this survey are to characterize species composition, relative abundance, and size structure. The following report is a brief summary of all activities conducted, general status of fish populations and future management options.

Acres: 112.3
Lake Type: Seepage
Regulations: Statewide Default Regulations
Shoreline Miles: 1.77
Public Access: 2 Public Landings
Maximum Depth (feet): 13

Wisconsin DNR Contact Info.

Al Niebur—Fisheries Biologist
Elliot Hoffman - Fisheries Technician

Wisconsin Dept. of Natural Resources
647 Lakeland Rd.
Shawano, WI 54166

Al Niebur Phone: 715-526-4227
E-mail: alan.niebur@wisconsin.gov

A copy of this report can be found online at:
http://dnr.wi.gov/topic/fishing/reports/

Survey Information

Site location	Survey Date	Water Temp. (F)	Target Species	Total Miles Shocked	No. of Stations	Gear	Dippers
Hatch Lake	6/4/14	75.0	All	2.23	5	Boomshocker	2

Survey Method

- Hatch Lake was sampled according to spring electrofishing (SEII) protocols as outlined in the statewide lake assessment plan. The primary objective for this sampling period is to count and measure adult bass and panfish. Other gamefish may be sampled but are considered by-catch as part of this survey.
- The entire shoreline was sampled with a boomshocker. All fish captured were identified to species and measured for length. A subsample of fish were weighed with age structures (otoliths) collected for age and growth analysis.
- Fish metrics used to describe fish populations include catch per unit effort, proportional stock density, length frequency distribution, and mean age at length.

Fish Metric Descriptions

CPUE, PSD, LFD and Growth

Catch per effort (CPUE) is an index used to measure fish population relative abundance which simply refers to the number of fish captured per unit of distance or time. For lake surveys we typically quantify CPUE by the number and size of fish per mile of shoreline. CPUE indexes are compared to statewide data by percentiles. For example, if a CPUE is in the 90th percentile, it is higher than 90% of the other CPUEs in the state.

Proportional stock density (PSD) is an index used to describe size structure of fish. It is calculated by dividing he number of quality size fish by the number of stock size fish for a given species. PSD values in the 30 to 50 percent range generally describe a balanced fish population.

Length frequency distribution (LFD) is a graphical representation of the percentage of fish captured by one inch size intervals. Smaller fish (or younger age classes) may not always be represented in the length frequency due to different habitat usage or gear sampling limitations.

Mean age at length is an index used to assess fish growth. Growth structures (otoliths, scales, or spines) are collected from a specified length bin of interest (e.g. 7.0-7.5 inches for bluegill). Mean age is compared to statewide data by percentile with growth characterized by the following benchmarks: slow (<33rd percentile); moderate (33rd to 66th percentile); and fast (>66th percentile).

Abundance Metrics

Species	Total CPUE (no per mile)	Percentile Rank	Overall Abundance Rating	Length Index (inches)	Length Index CPUE (no per mile)	Percentile Rank	Abundance Rating
BLUEGILL	124.2	57th	Moderate	≥ 7.0	9	55th	Moderate
LARGEMOUTH BASS	8.1	26th	Low	≥ 14.0	1.3	24th	Low
PUMPKINSEED	20.2	71st	Moderate - High	≥ 7.0	3.6	83rd	Moderate - High
YELLOW PERCH	11.7	53rd	Moderate	≥ 8.0	0	0	Low

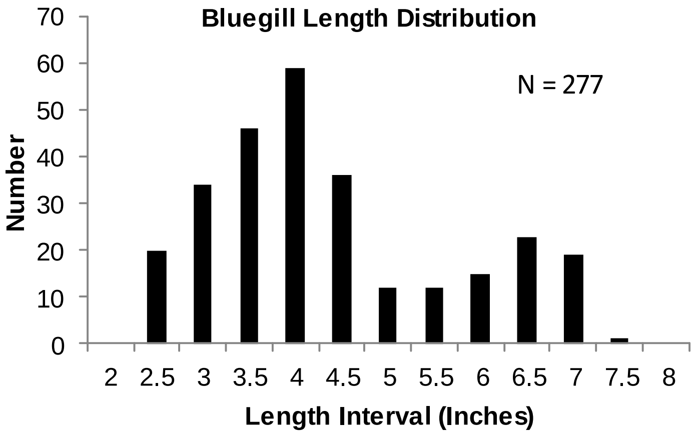
Size Structure Metrics

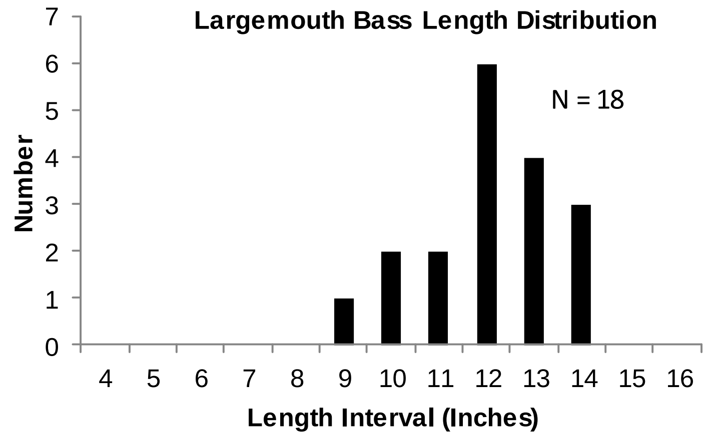
Species	Total	Average Length and (Range)	Stock and Quality Size (inches)	Stock No.	Quality No.	PSD	Percentile Rank	Size Rating
BLUEGILL	277	4.6 (2.5-7.7)	3.0 and 6.0	257	58	23%	32nd	Low
LARGEMOUTH BASS	18	12.4 (10.0 -14.3)	8.0 and 12.0	18	13	72%	69th	Moderate-High
PUMPKINSEED	45	5.2 (2.9 - 7.9)	3.0 and 6.0	43	17	40%	52nd	Moderate
YELLOW PERCH	26	4.9 (3.0 - 5.7)	5.0 and 8.0	15	0	0%	-	Low

2014 Spring Electrofishing (SEII) Summary Report

Hatch Lake (WBIC 282800)

Waupaca County





Stocking History					
Species	Year	Source	Age	Mean Length (inches)	Number Stocked
WALLEYE	2005	Private - Lake Assoc.	Large Fingerling	6.5	500
WHITE SUCKER	2005	Private - Lake Assoc.	Large Fingerling	6.5	3500
WALLEYE	2006	Private - Lake Assoc.	Large Fingerling	6.0	647
WALLEYE	2007	Private - Lake Assoc.	Large Fingerling	5.5	646
WALLEYE	2008	Private - Lake Assoc.	Large Fingerling	7.0	500
WALLEYE	2009	Private - Lake Assoc.	Large Fingerling	4.75	2400
WALLEYE	2013	Private - Lake Assoc.	Large Fingerling	6.0	490

Growth Metrics					
Species	Age Sample No.	Length Bin	Mean Age and Range (inches)	Percentile Rank	Growth Rating
BLUEGILL	5	6.0-6.5	6.6 (6-8)	<33rd	Slow
BLUEGILL	7	7.0-7.5	6.4 (6-8)	<33rd	Slow
LARGEMOUTH BASS	3	14.0-14.5	10.3 (8-12)	<33rd	Slow

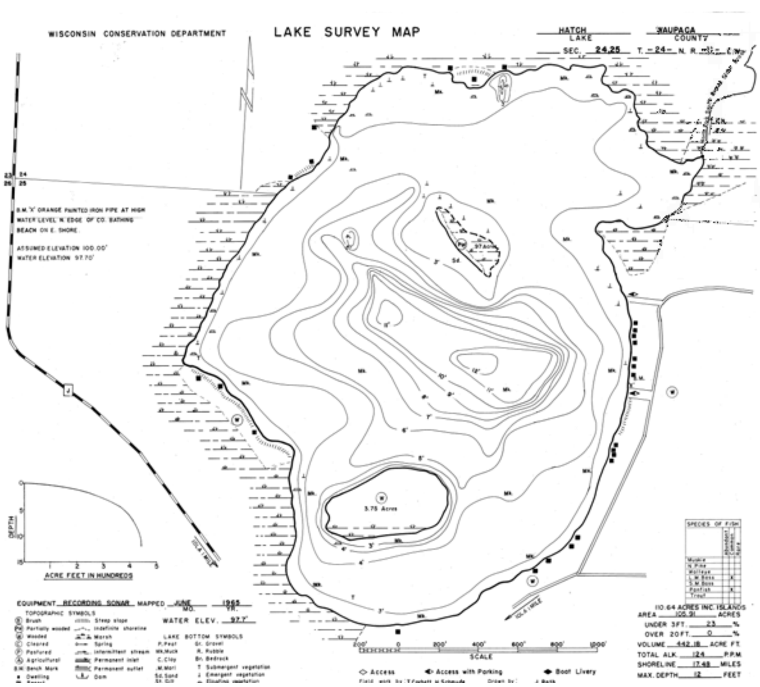
Summary

- Largemouth bass populations were at low density but showed above average size structure with proportions of 12.0+ inch bass at 72%. Growth metrics indicated slow growth with bass reaching legal size in about 10 years.
- Bluegill and pumpkinseed relative abundance was at moderate levels but the proportion of 6.0+ inch fish was low when compared to other lakes statewide. Growth metrics for bluegill indicated very slow growth.
- Other species sampled in low abundance included northern pike (1), walleye (1), yellow bullhead (4), black bullhead (1), rockbass (5), and black crappie (4).
- Walleye and northern pike were sampled in low number, however, our gear and sampling timeframe are not suitable to target these species.

Management Options

Management options for Hatch Lake should focus on preservation of habitat and water quality. The aerator operated by the lake association is an important management tool to minimize winterkill and should be maintained. Panfish (prey) populations are slow growing and management of predators may be the best option to improve size structure. Stocking predator gamefish and/or higher minimum length limits to increase predator population density would likely be the best option to improve size structure. Regulatory options would need to be vetted though a public input process. Tentative objectives are as follows:

- Increase bluegill PSD to 30-40%.
- Increase largemouth CPUE to 15-20 bass per mile.
- Initiate stocking quota of northern pike and/or largemouth bass and explore feasibility of increasing gamefish minimum length limits.
- Continue fisheries assessments on 8 year rotation (next survey in 2022).



F

APPENDIX F

Wisconsin Department of Natural Resources Comments

Comments to Hatch Lake Comprehensive Management Plan

WDNR Official Comments: Ted Johnson (Water Resources Management Specialist)

Comment Key:
Responses in blue by Tim Hoyman (Onterra, LLC)

Hi Tim,

I've looked over the plan and have a couple of comments.

1. Please change my phone number to (920) 362-0181
   Change has been made.

2. For watershed work, I recommend that you list the Waupaca County LCD as the first and primary contact. The DNR is certainly involved but I think that the County LCD is the best place to start.
   The management action is an educational initiative and lists the WDNR as a possible source of information along with the county, and UW-Extension.

3. EWM management. I'm a little confused as to what the expectations are for ProcellaCOR. I would hope that the expectation would be better than 2-years of control (including year of treatment). I realize that we do not know what the outcome of the last treatment will be but two years seems like, other than for nuisance control, a bit of a failure to me. When you say that past experience leads you to believe that this control strategy will last for about two years what is meant by that statement? A return to pre-treatment levels? If this is the case, and the lake returns to pretreatment levels in two years, would you recommend ProcellaCOR to be used again?
   As described in the action's description, the at least two-year expectation was based upon monitoring of lakes that had completed similar treatments. We only had two years of data to access when the OFD was written in spring 2022. We now have three years of data and have updated the text in the action to reflect that. Additional wording has been included to explain that it would slowly come back to near or actual pretreatment levels over time.

4. I like the proposed monitoring strategy.
   Thank you.

Thanks,

Ted

G

APPENDIX G

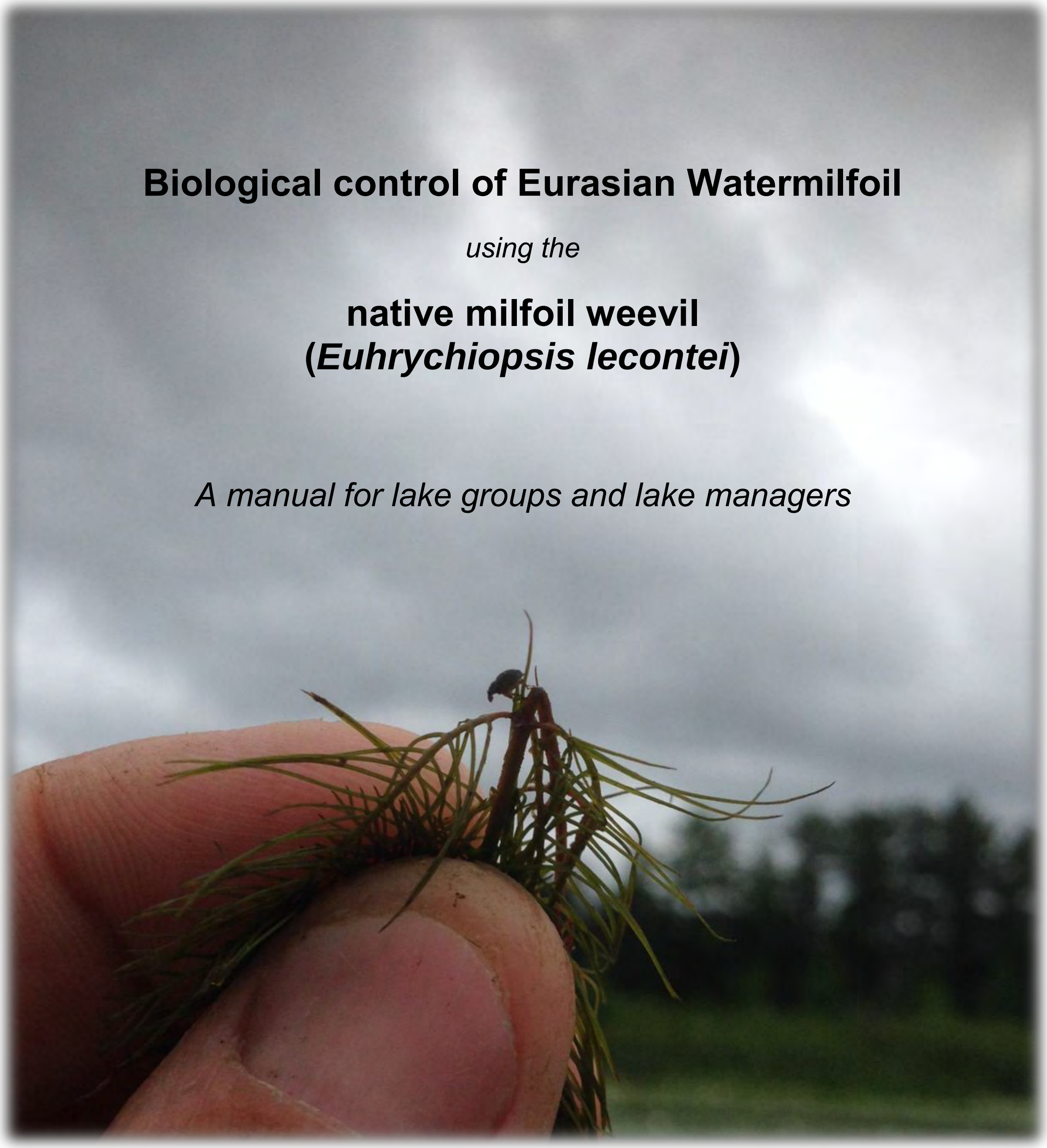
Milfoil Weevil Studies & Reports from Golden Sands RC&D

Biological control of Eurasian Watermilfoil

using the

native milfoil weevil
(*Euhrychiopsis lecontei*)

A manual for lake groups and lake managers



June 2017

Golden Sands
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& Development Council, Inc.

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Acknowledgements

This manual, and the mass rearing pilot study that produced the rearing content, was made possible through grant funding from the Wisconsin Department of Natural Resource (WDNR) Aquatic Invasive Species Program: ACEI-123-12.

This manual was written by Amy Thorstenson, M.S., Executive Director and Regional Aquatic Invasive Species Coordinator for Golden Sands Resource Conservation & Development Council, Inc. Thorstenson has 13 years of experience with milfoil weevil rearing and research, including her thesis work on shoreline habitat requirements of milfoil weevils. You can find her published work in the Journal of Aquatic Plant Management. (See literature cited section for full citation.)

Additional technical assistance came from Paul Skawinski, Dr. Ronald Crunkilton, and Jodi Lepsch. Paul Skawinski, M.S., is formerly the Regional Aquatic Invasive Species Education Specialist for Golden Sands RC&D, Inc., and currently with University of Wisconsin-Extension Lakes Program. Skawinski has 11 years of experience in milfoil weevil rearing and research, including his thesis work on the effects of in-lake and shoreland variables on milfoil weevils. Dr. Ronald Crunkilton, Ph.D., has been a fisheries and water resources professor with the University of Wisconsin-Stevens Point (UWSP) since 1990. He has been advising the graduate research on biological control of Eurasian watermilfoil in collaboration with WDNR since 2008, including Thorstenson's and Skawinski's research. Jodi Lepsch, WDNR Water Resource Management Specialist, has been heavily involved in monitoring weevil study lakes since 2011. She is currently compiling this data for pier review.

Monitoring data was collected and analyzed as a collaborative effort with Jodi Lepsch, WDNR, and staff from Beaver Creek Reserve Citizen Science Center.

Many photos are used with permission from Paul Skawinski, author *Aquatic Plants of the Upper Midwest*. Many thanks to Paul for so many excellent close-ups of both weevils and milfoil that show such critical detail.

The mass rearing pilot study would not have been possible without the participation of several rearing teams: Goose Lake Association and team leader Reesa Evans of Adams County Land Conservation Department; Lake Holcombe Improvement Association and team leader Dr. James "Doc" Dougherty; Swift Nature Camp, Minong Flowage Association, Shallow Lake volunteers, and team leader Dave Blumer of SEH, Inc.; St. Croix County Parks Department, Perch Lake volunteers, Lake Hallie volunteers, and team leaders at the Beaver Creek Reserve Citizen Science Center. Without their hard work and dedication to feeding and caring for their "weevil babies", we would not have been successful in developing a rearing method for volunteers.

I. INTRODUCTION

Eurasian watermilfoil (*Myriophyllum spicatum* L.) is a non-native aquatic plant from Eurasia that aggressively invades littoral zones of lakes. Introduced to the United States in the 1940's (Couch and Nelson 1986), it is now found in 45 states and four Canadian provinces (USDA, NRCS 2010). By the end of 2010, 539 waterbodies in Wisconsin had confirmed occurrences of Eurasian watermilfoil (WDNR 2011). The cumulative effect of Eurasian watermilfoil impacts lake ecology, decreases recreational, sporting and aesthetic values of the waterbodies, and decreased property values (Newroth 1985). The magnitude of the problem is so large that several million dollars are spent annually on Eurasian watermilfoil control in the northern tier states (Mullin et al. 2000).

Historically, control options for Eurasian watermilfoil have relied heavily on mechanical harvesting or chemical treatments, which do not provide a long term solution since they require repeated application (Crowell et al. 1994, Getsinger et al. 1997, Parsons et al. 2001). Concerns regarding the potential hazards posed by putting toxic herbicidal chemicals into our public waterways have been expressed by resource managers since Eurasian watermilfoil first emerged as a problem (Blakey 1966). Research examining the effects of herbicides and insecticides at low, residual levels that now commonly contaminate aquatic communities has been limited. Recently, Relyea (2009) found that even residual levels of some pesticides (diazinon, endosulfan) resulted in 24-84% mortality in leopard frogs (*Rana pipiens*), and that mixtures of chemical residuals may be much more toxic (99% mortality in leopard frogs) than the individual chemicals. Moreover, additional concerns regarding chemical use have arisen due to the recent development of flouridone resistance in several biotypes of hydrilla, spurring renewed interest in alternatives to chemical controls (Michel et al. 2004, Netherland et al. 2005).

Declines in Eurasian watermilfoil have been associated with several herbivorous insects: a naturalized moth, *Acentria ephemerella* (Denis & Schiffermüller), a native midge, *Cricotopus myriophylli* (Olivier), and the native milfoil weevil, *Euhrychiopsis lecontei* (Dietz) (Painter and McCabe 1988, Kangasniemi et al. 1993, Julien and Griffiths 1999). Primary focus for biological control has been on the latter (Sheldon and Creed 1995, Newman et al. 1996, Buckingham 1998, Newman 2004, Newman et al. 2006). Research suggests this milfoil weevil has potential to biologically control Eurasian watermilfoil, but more study on factors limiting populations adequate for control is necessary (Creed and Sheldon 1995, Sheldon and Creed 1995, Creed 2000, Jester et al. 2000, Madsen et al. 2000, Newman 2004, Cuda et al. 2008, Reeves et al. 2008).

Biology of Eurasian watermilfoil

Eurasian watermilfoil (*Myriophyllum spicatum*) has spread to waterbodies across the U.S. by boaters, recreationalists, and various aquatic industries. Once introduced, Eurasian watermilfoil spreads rapidly via fragmentation (Nichols 1975). This submersed aquatic plant goes through two flowering periods each summer, after which it fragments into pieces. Subsequently, each fragment may sprout roots and can remain afloat and stay viable for several weeks until it drifts to a suitable site, where it can take root and become another plant (Kimbel 1982, Rawson 1985). As a perennial plant, the lower portions of the stems may remain green during the winter (Reed 1977, Kimbel 1982), allowing the plant to start growing and become well established by April, much sooner than native aquatic plants

(Aiken et al. 1979). Then, it grows rapidly, reaching the water surface and then spreading into a dense, tangled canopy, shading out other aquatic plants (Aiken et al. 1979).

The dense canopy of Eurasian watermilfoil can alter the physiological and chemical characteristics of littoral zones. It increases dissolved oxygen, carbon dioxide, and pH fluctuations, inhibits water circulation, and promotes localized temperature stratification (Carpenter and Lodge 1986, Engel 1994). Eurasian watermilfoil can aggressively out-compete the native aquatic plants, which rapidly decreases the diversity of the lake's plant community (Aiken et al. 1979), which in turn can alter fish communities (Crowder and Cooper 1982, Savino and Stein 1982, Diehl 1988, Dionne and Folt 1991). The tangled canopy at the water surface can become dense enough to hamper recreational activities, clog water intake pipes, and create a stagnant breeding ground for mosquitoes ((Aiken et al.1979, Bates et al. 1985, Newroth 1985).

Biology of the milfoil weevil

The aquatic milfoil weevil, *Euhrychiopsis lecontei*, is native to North America, is broadly distributed across Wisconsin (Jester et al. 2000), and its lifecycle holds the key to its potential as a biological control for Eurasian watermilfoil. The adult weevil spends the winter hibernating on-shore at the soil-leaf litter interface (Newman et al. 2001). After ice-out, adults move out to milfoil beds to feed on apical stems and begin to lay eggs once water temperature reaches 15°C (May-June) (Newman et al. 2001). In spring, adult flight muscles are well-developed, and they have been documented to fly in spring (back to the milfoil beds), but in summer, flight muscles are atrophied while energy is re-allocated to reproduction (Newman et al. 2001). Females on average lay two to four eggs per day, and may lay multiple eggs on one meristem (Sheldon and O'Bryan 1996a, Sheldon and Jones 2001). Larvae eat the meristem then bore into the stem to feed, mature and pupate (Newman et al. 1996). They mine (i.e. eat) an average total of 15 cm of stem tissue (Mazzei et al. 1999). Weevils normally pupate within the stem approximately 50 to 75 cm from the meristem, later emerging as adults (Mazzei et al. 1999). They spend little time outside of the stem until they are adults.

At typical summer lake temperatures of 25°C, the full life cycle can be completed within 21 days, and 3-5 generations may be produced per summer (Mazzei et al. 1999). At the optimal developmental temperature of 29°C (as in controlled laboratory situations) the full life cycle, egg to adult, takes 17 days (Mazzei et al. 1999). Theoretically, the cycle can be shortened in an artificial rearing situation where temperatures are maintained closer to optimum.

In fall (September through November), weevils move to shore where they overwinter at the soil-leaf litter interface (Newman et al. 2001). In spring, between ice-out and mid-May, they return to the lake, where they affect milfoil (Newman et al. 2001). It is currently unknown how they move to and from shore. They have been documented to fly in spring, but this has not been documented in fall (Newman et al. 2001). It is unknown whether they are strong enough fliers to select habitat, their direction is controlled by wind speed and direction, or they may simply raft to shore in fall on milfoil fragments.

The milfoil weevil's potential as a biological control agent

Native insects are preferred for use in biological control of invasive species due to the reduced risk of impacts to native, non-target plants, especially agricultural crops. Studies on several native or naturalized insects for controlling Eurasian watermilfoil have determined them to be poor candidates for use in biological control because they were: 1) too general in their feeding preferences [e.g. the moth *Acentria ephemerella (*Dennis and Schiffermuller; = *A. nivea* Olivier; = *Acentropus niveus* Olivier); Batra 1977, Buckingham and Ross 1981], 2) incapable of providing control (e.g. *Phytobius leucogaster* (= *Litodactylus leucogaster* Marsham); Buckingham et al. 1981], or 3) too difficult to rear to the high population densities needed (e.g. the milfoil midge *Cricotopus myriophylli* Olivier; Kangasniemi et al. 1993). In contrast, evaluations of studies on *E. lecontei* (hereafter referred to as the milfoil weevil) have found it to be suitable on all three aspects (Sheldon and Creed 1995, Newman 2004).

The milfoil weevil has demonstrated a preference for Eurasian watermilfoil, even when native milfoil species are present, and is not known to cause damage to other aquatic macrophytes (Solarz and Newman 2001). One reason may be that Eurasian watermilfoil may lack the specific plant defenses that native milfoils possess from coevolving with milfoil weevils, which would give the weevil an advantage against its exotic host (Newman 2004). Adults initially visually target plants with the correct host-plant shape (Reeves et al. 2009), and then respond to the chemical attractants (glycerol and uracil) that are produced at higher concentrations by Eurasian watermilfoil than native milfoil species (Marko et al. 2005).

Control of Eurasian watermilfoil by milfoil weevils is achieved by larval stem-mining, which causes loss of buoyancy, nutrient depletion, and secondary infections. Stem-mining damages the vascular tissue (Newman et al. 1996) and releases cellular gases, which reduces stem buoyancy and causes the plant to sink below the water surface (Creed et al. 1992). This reduces the dense, tangled canopy at the water surface that causes most ecological and public recreation impacts (Sheldon and Creed 1995). Larval stem-mining also reduces the transfer of nutrients and carbohydrates from leaves to stems to roots (Newman et al.1996). Larvae also create openings for secondary infections by pathogens and deposit frass (waste) in the stem, which may promote those infections (Creed 2000).

Studies have shown the milfoil weevil performs better on Eurasian watermilfoil than on native *Myriophyllum* species. Females will lay over four times as many eggs on Eurasian watermilfoil as on northern watermilfoil (*M. sibiricum*) (Sheldon and Creed 1995). Juveniles exhibit faster developmental rates (1-3 days), higher survival rates, and adults emerge from pupal chambers having higher mass than those reared on northern watermilfoil (Newman et al. 1997, Solarz and Newman 2001). The nutritional quality of Eurasian watermilfoil versus native milfoils may play a significant role in this difference, but this conclusion lacks adequate study (Newman 2004).

Milfoil weevil-related declines

High-density beds of Eurasian watermilfoil in some lakes have exhibited periods of rapid decline in association with the milfoil weevil (Creed and Sheldon 1995, Sheldon 1997, Creed 1998, Lillie 2000), including ten lakes in Vermont (Madsen et al. 2000). Due to a lack of pre-decline data, however, the reasons for these seemingly natural population collapses have generally not been well documented. One of the few well-documented studies of a natural decline of Eurasian watermilfoil reported a reduction from 123 g dry matter/m^2 to 23, 5, 44 and 12, g dm/m^2 in subsequent samples over a 3-year period in a 12 ha man-made lake in Minnesota (Newman and Biesboer 2000). In this system, densities of the weevil were the highest yet reported for Minnesota lakes at 103 weevils per m^2 (1.6 to 2.0 weevils per milfoil stem) at start of the study, when milfoil density was greatest (Newman and Biesboer 2000).

Stocking milfoil weevils in controlled laboratory and field enclosures have shown the herbivore is capable of controlling Eurasian watermilfoil (Creed and Sheldon 1995, Sheldon and Creed 1995, Newman et al. 1996). However, open field trials have shown mixed results. In one supplemental stocking study in Wisconsin, Jester et al. (2000) associated significant within-season declines in Eurasian watermilfoil study plots with weevil densities, ranging from 0.5 to 3 per stem, in six out of 12 treatment lakes. These were open-plot stocking trials and Jester et al. (2000) theorized that one possible reason the other six plots did not reach control-level populations was that the weevils that were stocked may have simply left from the study plots.

Supplemental stocking experiments have been conducted in association with the mass rearing study that produced this manual. Monitoring surveys have been conducted annually by the WDNR to track the progress of Eurasian watermilfoil and the milfoil weevils in Pine Lake (an embayment of Lake Holcombe, Chippewa County) and Perch Lake (St. Croix County). The following excerpt was contributed by the WDNR as a summary of results to date on these two study lakes. A manuscript for peer review is in production. Monitoring of Perch Lake and Lake Holcombe is ongoing. Fluctuations in both the milfoil and the weevil populations are expected. Little is known about the long-term effects of weevil stocking, making these two lakes important case studies to follow.

1) <u>Pine Lake</u>

   Pine Lake was surveyed for weevil presence in 2011 and it was found to be lacking a natural weevil population. Lake Holcombe Improvement Association stocked weevils into Pine Lake in 2011, 2012 and 2013. Over 10,000 weevils were produced through mass rearing by volunteers (Section V) and released over the course of the stocking. The aquatic plant community and weevil population were surveyed annually 2011-2016 in June (spring) and August (summer) following the monitoring for professionals method found in Section IV. Figure 1 shows the frequency of EWM during the spring and summer surveys. Though results were variable, there is a clear linear trend of decreasing EWM frequency over time. Figure 2 shows the frequency of weevils and the frequency of characteristic feeding damage (evidence) at sites with EWM. It is apparent that the weevil population successfully overwinters and can locate EWM throughout the lake. Figure 3 shows the total number of individual weevils that were found and the average number of weevils per stem. This shows that even as EWM declines the weevil population is reproducing and can sustain the population throughout the growing season.

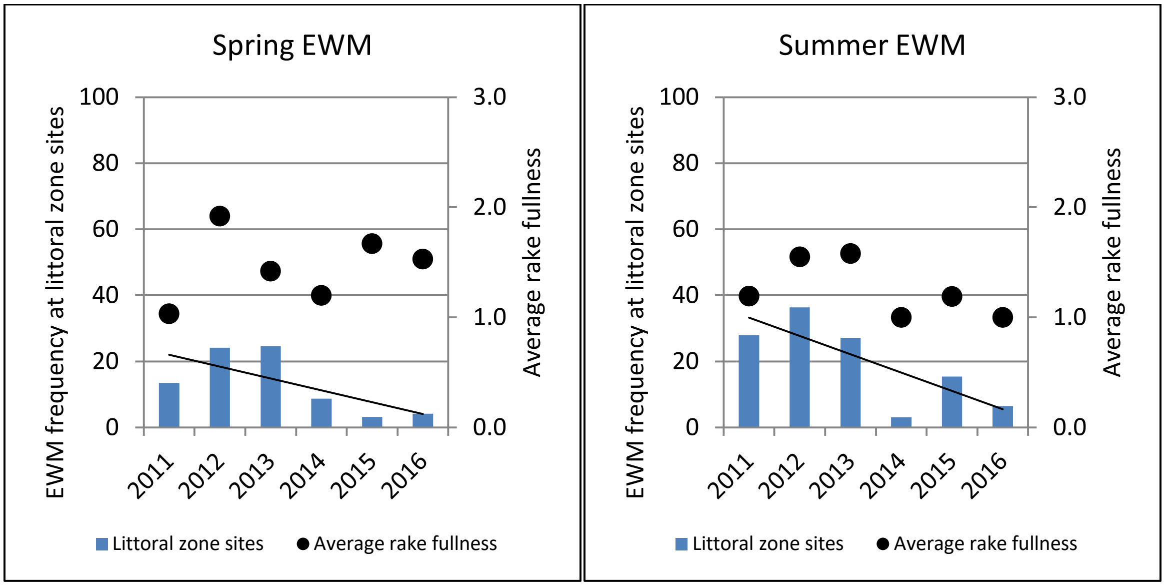


Figure 1. Frequency and average rake fullness of EWM at littoral zone sites in Pine Lake.

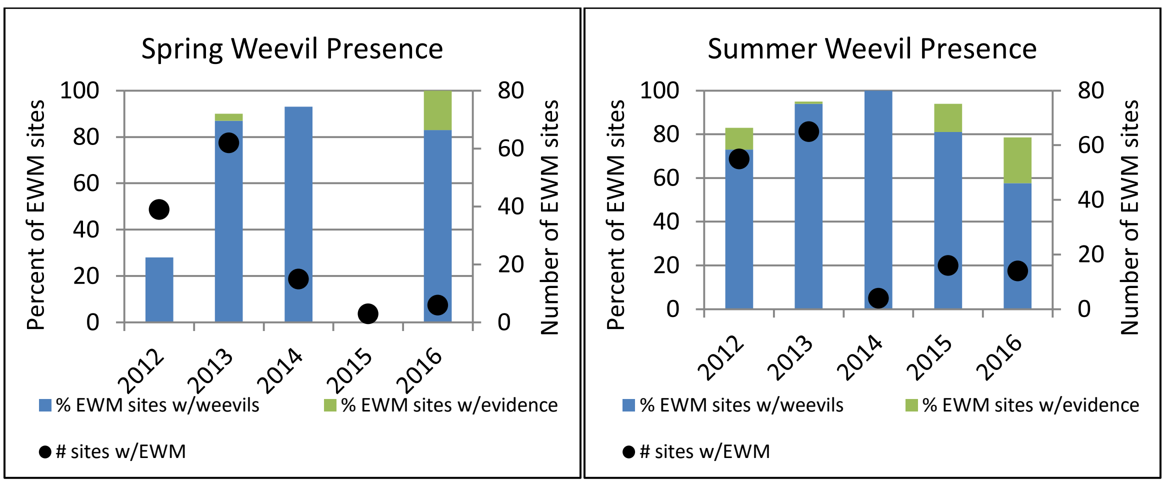


Figure 2. Frequency of weevil presence at EWM sites in Pine Lake.

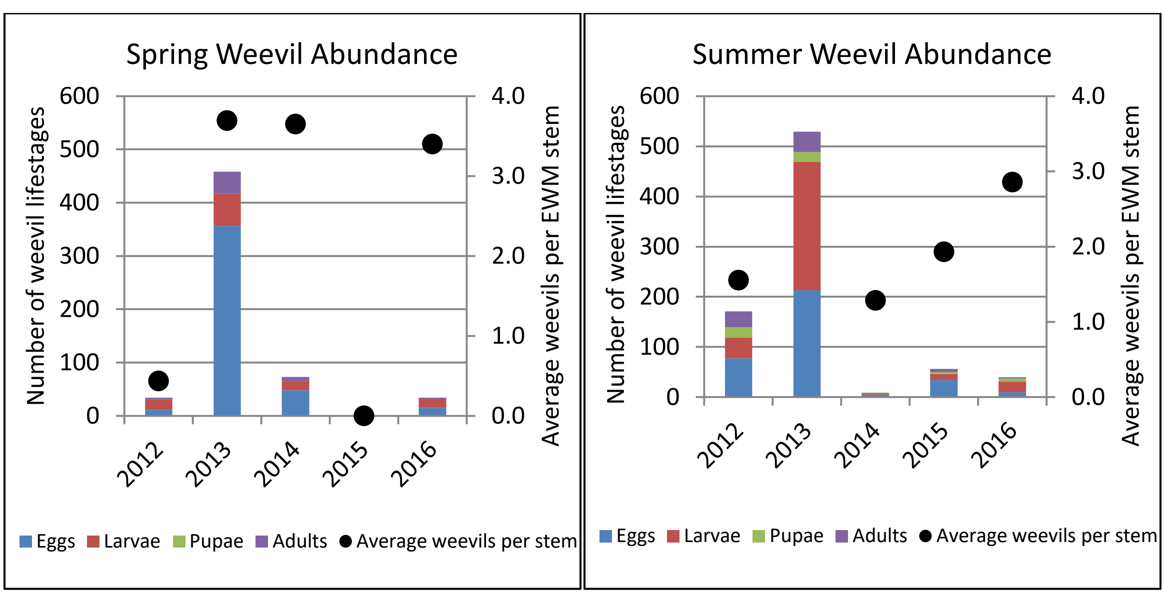


Figure 3. Total number of individual weevils and average number of weevils per stem in Pine Lake.

2) <u>Perch Lake</u>

Beaver Creek Reserve – Citizen Science Center stocked weevils into Perch Lake in 2012 and 2013. Over 21,875 weevils in total were raised through mass rearing by volunteers (Section V) and stocked. The Perch Lake aquatic plant community was surveyed annually 2012-2016 in July using the standardized point intercept method. The weevil population was surveyed in 2012, 2013 and 2014 following the monitoring for volunteers method found in Section IV. The weevil population was monitored in 2016 following the monitoring for professionals method. Figure 4 shows the frequency of EWM. After a significant drop in EWM during 2014, the population has been on a steady increase. That being said, EWM levels remain below pre-stocking levels of 2012 and the linear trend shows a decrease over time. Figure 5 shows the frequency of weevils and the frequency of characteristic feeding damage (evidence) at sites with EWM. The Perch Lake weevil population also successfully overwinters and can locate EWM throughout the lake. Figure 6 shows the total number of individual weevils that were found and the average number of weevils per stem. This shows that the weevil population is reproducing but not as abundantly as is seen in Pine Lake. The population continues to sustain itself after stocking activities have ceased.

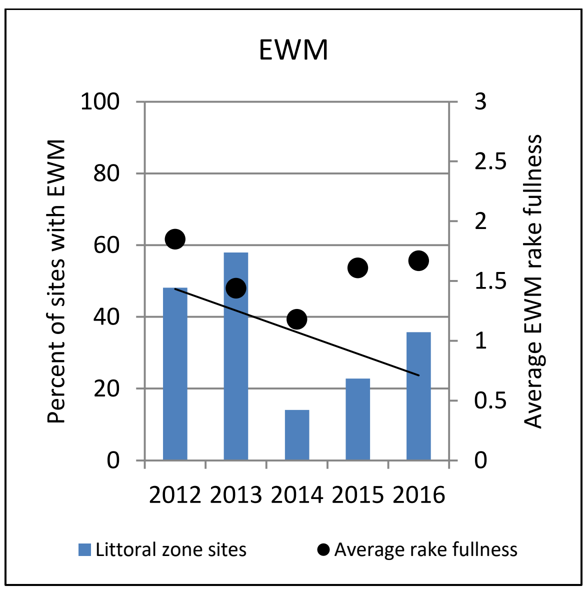


Figure 4. Frequency and average rake fullness of EWM at littoral zone sites in Perch Lake.

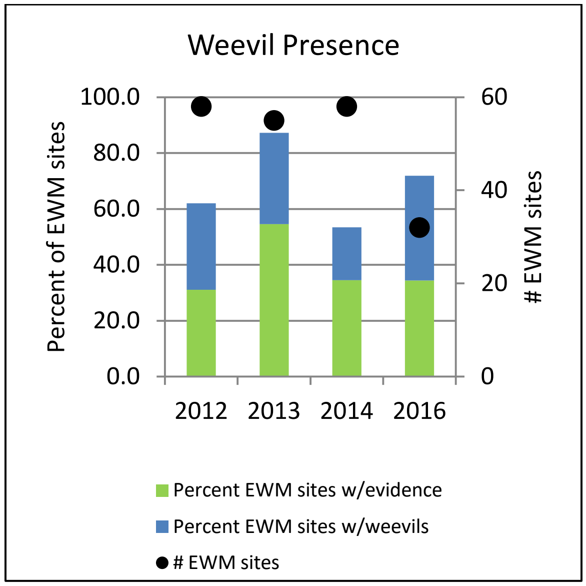


Figure 5. Frequency of weevil presence at EWM sites in Perch Lake.

Currently, there is no prescription for predicting whether the milfoil weevil will be able to reach control levels in a given lake, or how long it will take to reach control levels. There is a need for more long-term studies of weevil stocking programs.

Lake characteristics affecting weevil success

Shoreline habitat for overwintering may be one important factor in sustaining high milfoil weevil populations. In fall (September through November), weevils move to shore where they overwinter at the soil-leaf litter interface (Newman et al. 2001). Newman et al. (2001) found that populations were most commonly found at two to six meters from the shoreline, and were significantly lower in sites with soil moisture >15%. Thorstenson et al. (2013) concurred with these findings. Jester (1998) found milfoil weevil population density correlated positively with natural shoreline vegetation, and negatively with bare, sand shorelines, indicating that human disturbance can be a limiting factor for sustaining weevil populations. Newman et al. (2001) found that weevils can be successful on natural grass riparian areas (i.e., prairie sites). Thorstenson et al. (2013) found no conclusive correlation between weevils and the *type* of leaf litter present (i.e. pine needles, deciduous tree leaves, grasses, or herbaceous litter), but did find that *depth* of leaf litter was correlated with weevil occurrence, suggesting that activities that remove leaf litter (mowing, raking) may be disadvantageous to weevil populations.

Weevil success may also be limited by predation. Adult weevils are more vulnerable to predation than the larval and pupal life stages that are concealed inside the milfoil stems. The longevity of the egg-laying adult females are critical to population growth (Ward 2002). Modeling suggests that increasing an adult female's lifespan from five to 10 days can result in an 8-fold increase in end-of-summer population densities (Ward 2002). Studies on predation by vertebrates have found that while yellow perch (*Perca flavescens*) do not appear to feed on weevils (Creed 2000), sunfish and bluegills (*Lepomis sp.*) do and could limit milfoil weevils from reaching densities capable of suppressing Eurasian watermilfoil

(Ward and Newman 2006). Ward and Newman 2006 found that sunfish catch rates greater than 25-30 sunfish per 24 hr trapnet, as in the case of stunted populations, may result in relatively low weevil densities (<0.1/stem). Because dense Eurasian watermilfoil beds may produce stunted sunfish populations, this may perpetuate the Eurasian watermilfoil problem by increasing predation on milfoil weevils (Engel 1995). To break this cycle, more study on the potential to better understand and ameliorate sunfish predation pressure on weevils is needed.

Predation on weevils by invertebrates is less well studied. Ward and Newman (2006) cite two studies that suggest milfoil weevils do not appear to be vulnerable to most invertebrate predators. In contrast, Tamayo (2003) found a negative correlation between weevils and Hirudinoidea (leeches) and Hydrachnida (water mites) densities, suggesting a need for more study on invertebrate predators and competitors.

For the most part, general lake characteristics appear not to influence the distribution of presence and abundance of weevils. When measured at the whole-lake level, temperature, dissolved oxygen, pH, water clarity, nitrogen, chlorophyll a, alkalinity, and conductivity showed no correlation with milfoil weevil densities (Jester et al. 2000, Skawinski 2014). However, because Eurasian watermilfoil is known to alter in-bed pH, dissolved oxygen, carbon dioxide and temperature circulation (Engel 1994), bed-level parameters may prove to be a factor in weevil densities. The sediment a milfoil bed grows in was found to be an important factor in the success of weevils; Skawinski 2014 found weevils were negatively correlated with coarseness of substrate, meaning that weevil densities were higher in EWM beds with finer, muckier substrates. The size and depth of a milfoil bed are also important; Jester et al. 2000 found higher densities of weevils in larger beds in shallower water. Creed 2000). Jester et al. (2000), Reeves et al. (2008), and Creed (2000) all call for further studies on bed-level conditions that may affect milfoil weevil populations.

Integrated use

A relatively new area of exploration in biological control is integrated use, or the coordinated use of multiple control methods. For example, researchers in water hyacinth control have found success by combining one biological control agent with a second agent, or with limited herbicide applications (Van 1988, Haag and Habeck 1991). Combining milfoil weevils with a second biological control agent may also hold promise. Shearer (2009) found that the endophytic fungus *Mycoleptodiscus terrestris* was only detrimental to Eurasian watermilfoil when the plant was stressed, and suggested that milfoil weevils may be useful in creating that stress. Although Skawinski 2014 found no correlation between milfoil densities and the presence of weevils and *M. terrestris*, looking back at the mixed results with stocking weevils (Sheldon 1997, Madsen et al. 2000, Reeves et al, 2008), perhaps one of the differences between success or failure was dependent on the presence of a second, unknown agent. The question remains whether there may be a second biological control agent we have yet to identify that, while alone it makes no significant impact on milfoil, when paired with weevils it can be the critical stressor. This question seems worthy of further study.

Experimentation with carefully coordinated integrations, using targeted applications of mechanical controls, may also hold promise (Newman and Inglis 2009, Sheldon and O'Bryan 1996b). For instance, broad-scale applications of mechanical controls appear incompatible with weevils, since weevils lay their eggs on the tops of milfoil plants and mechanical harvesting, by design, removes the tops of plants. However, mechanical harvesting of less than 15% of the milfoil beds may avoid the detrimental impacts to weevil populations and allow the strategic use of both control methods together (Newman and Inglis 2009).

Although broad-scale use of chemical herbicides is incompatible with milfoil weevils because it removes the food base weevils need to survive, targeted use of the two control methods in separate areas of the same lake may be hold potential for control. In an unpublished report on Bass Lake in St. Croix County, WI, Jester (2000) found that weevil densities in untreated beds adjacent to chemically treated beds were slightly higher than that of control beds that were far from the treatment areas (0.800 weevils per stem versus 0.617 weevils per stem, respectively). This difference was not significant, but may have been attributable to adult weevils emigrating from treated beds into adjacent beds, or may have been due to the fact that treated beds were usually closer to shore where weevils tend to concentrate (Jester 2000). Additional studies are needed to understand the potential of integrating chemical and biological controls.

II. IDENTIFICATION OF MILFOIL WEEVILS

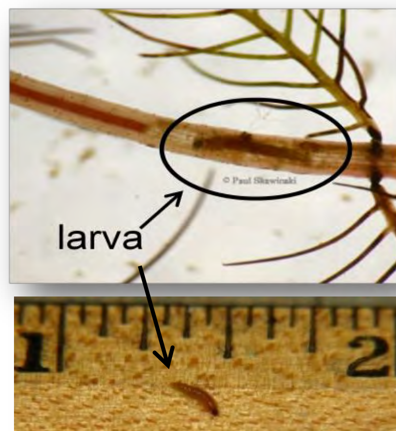
The native milfoil weevil, *Euhrychiopsis lecontei*, is a fully aquatic weevil that spends most of its life cycle under water. They are genus-specific feeders, meaning they feed only on milfoil species, therefore a weevil found on milfoil is likely to be *E. lecontei*. However, there are other native weevils that may be found on milfoil, as well as other beetles and mites that may have a generally-similar appearance. When monitoring for *E. lecontei* populations, it is important to learn to distinguish between these bugs.

Identification features of *E. lecontei*:

Eggs are small (0.5mm long), elliptical, and creamy-yellow in color. With strong magnification (50X), there is often visible a black or rust-colored squiggly string-like material wrapped around the egg. The female weevil lays her eggs on the apical meristem (growing bud) of the milfoil plant. They lay one egg at a time, but it is not unusual to find more than one egg on a meristem. Eggs hatch after 3-6 days.

Paul Skawinski

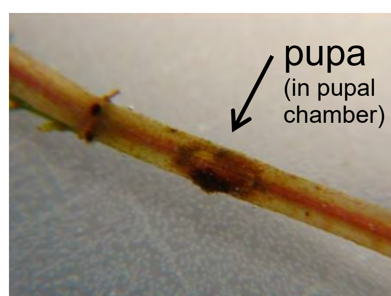
Larva are very tiny when they first hatch, but grow to a total length of 4.5 mm after 8-15 days. They are creamy-white in color, with a smooth, shiny, mahogany-black head capsule. Under 30X magnification, no mouth parts are visible, and they have no legs, tail, or any kind of hairs or bristles.



Paul Skawinski

Paul Skawinski

Pupa are in the transitional phase of the life stage, so the organism may more closely resemble the larva or the adult, depending on what stage of pupation the weevil is in. The pupal chamber is an oval-shaped hole completely contained within the stem. There is no outward bulging or blistering, and no cocoon casing made, although the entry hole is often covered with a black cotton-like material. The chamber will be located further down the stem than the larva, typically 0.25 - 0.5 m down from the tip. The pupal chamber appears as a darkened oval section of the stem. Larval tunneling will likely be visible just above the chamber, but not always. The pupal stage lasts about 9-12 days. If the weevil has already vacated the pupal chamber, the chamber will appear as a large oval hole, or crater, in the stem.



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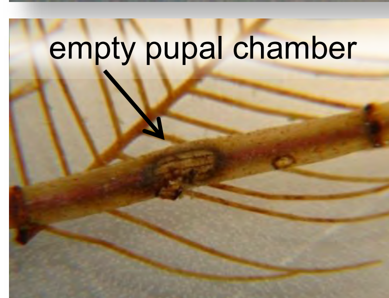

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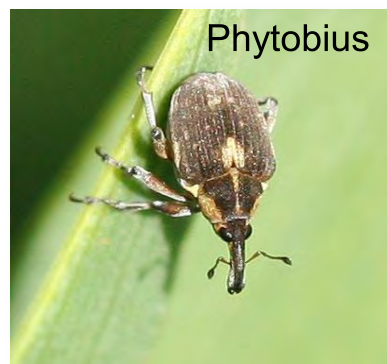
Adults feed on the milfoil leaves and will be found on the top 0.5 m of the stem, usually on or near the meristems, where the females lay their eggs. They are not capable of flight during the summer, but they may drop off of stems as they are picked, making it less likely to find adults than the juvenile stages. Adults are about the size of a sesame seed, just 2 - 3mm long, with a plump abdomen and a long snout (all weevils have them). They usually have distinctive striping on their backs, but the more definitive features are the raised bumps on the 7th ridge of the elytra (wing coverings) and long setae (hairs) on their legs. These are visible with 30X magnification. (See Appendix A for microphotography of these distinguishing characteristics.)

Feeding habits are a helpful way to distinguish *E. lecontei* from similar-looking weevils. Eggs are only laid on apical meristems, not on flowers. When they first hatch, the tiny larva will chew up the meristem, then burrow into the stem and mine the stem. They will exit and re-enter the stem several times before pupating, so it is possible to find them on the exterior of the stem. They will mine a total of about 15 cm of stem tissue as they make their way down the stem. The pupal chambers are fully contained within the stem, with no blistering bulge or cocoon formed.

Look-alikes in the aquatic realm are very few, but there are two aquatic weevils similar in appearance. *Bagous restrictus* is a non-species specific feeder that may be found on *M. spicatum* by happenstance. It is easy to tell *B. restrictus* apart from *E. lecontei* by simple appearance. (See Appendix A for photos and descriptions.)

More likely to be mistaken for *E. lecontei* is the nearly-identical *Phytobius leucogaster*. Like *E. lecontei*, Phytobius is also a native weevil and a genus-specific feeder of milfoils. Like *E. lecontei*, its entire reproductive cycle takes place on milfoil, and the eggs, larva, and pupa look identical. However, there are differences in where they are found on the plant; Phytobius cannot stay under water for more than 8 hours, therefore this weevil tends to keep to the emergent part of the milfoil plant – the flowering stalk. The eggs are laid on the flowering tips (the ovaries), not on apical meristems. The emerging larva chews up the flower ovaries, then chews up the flower stalk and may burrow into it, but is usually not found in the lower, submergent parts of the stem. It will then form a pupal chamber just below the flower on a part of the stem that is floating on the water surface, unlike *E. lecontei* pupa that are found much farther down on the submerged parts of the stem. The Phytobius pupal chamber is formed by building a cocoon on the outside of the stem that looks like a transparent blister the color of brown glass.



The adult Phytobius feed on milfoil leaves by stripping the leaves off the stem. Where there has been heavy Phytobius feeding, the milfoil stems may look like naked strands of spaghetti. Phytobius adults are capable of flight year round, which can make them harder to collect than *E. lecontei*. (See Appendix A for photos of indicative characteristics of Phytobius adults.)

Phytobius weevils may help to suppress Eurasian watermilfoil reproduction by reducing seed production to some degree. However, Phytobius does not appear to create enough feeding damage to suppress the overall health and vigor of the plant, and so is not viewed as a useful biological control agent. Because Phytobius inhabits different sections of the milfoil plant, it does not appear to compete with *E. lecontei* and would be unlikely to negatively impact a weevil stocking program.

III. USING BIOLOGICAL CONTROL

Evaluation criteria

Biological control is a good option for some lakes but not all. While the science of EWM management is ongoing, some good guidance has developed from existing studies. Considerations include:

1. Time committment - Biological control moves on "Mother Nature's" timescale. Natural crashes of milfoil have occurred after over 30 years of heavy Eurasian watermilfoil infestation, and investigations of these crashes have found *E. lecontei* present. Boom and bust relationships are common in predetor-prey population dynamics. If milfoil weevils are naturally present, milfoil may "go bust" naturally sometime in the future. The idea behind stocking is to make that happen in the immediate future. However, stocking should be looked at as a 3-5 year project, minimum, with annual monitoring to follow and track both the milfoil and the weevil populations. Because of it may take years to see the benefits of stocking weevils, biological control may not be right for lake groups looking for more immediate results.

2. Control ≠ Eradication - Lake groups chosing biological control would also need to accept that biological control would never eradicate Eurasian watermilfoil completely; it would only control it. Milfoil would still be present, but would be less dominant, allowing the native plants to rebound. Low levels of milfoil persisting in the lake are actually important in maintaining a weevil population for long term biological control. In Lake Holcombe, even when Eurasian watermilfoil could only be found at just 4 sample sites, the weevils persisted at 1.3 weevils/stem, showing that the weevil population was using whatever milfoil they could find to hang on in Lake Holcombe. When milfoil rebounded in 2015, showing up at 16 sample sites, the weevils rebounded as well (4.3 weevils/stem). These low level fluctuations in the predator and prey populations is what is typically expected in biological control.

3. Lake characteristics - Many lake characteristics have been studied. Many have no effect on weevil success, but several key factors do. (See Section I for a thorough discussion of this. See Figure 1 for a decision tree illustrating these how lake characteristics can help guide the decision to use biological control.) We know that weevils are more successful in lakes with:

    a) milfoil beds that are:
        - dense
        - widespread
        - growing in shallow water

- growing in fine sediments
- growing close to shore

b) natural shoreline habitat to overwinter in
c) balanced fish communities (no stunted sunfish populations)

Lake groups considering biological control cannot control where the milfoil beds are growing, but they do, as a collective, have control over the shoreline. Lake residents can choose to restore shoreland habitat to provide more hibernation sites for weevils. Additionally, mowed areas can be allowed to grow long after Labor Day and left unmowed and unraked until Memorial Day, to leave leaf litter and long grasses for weevils to hibernate under.

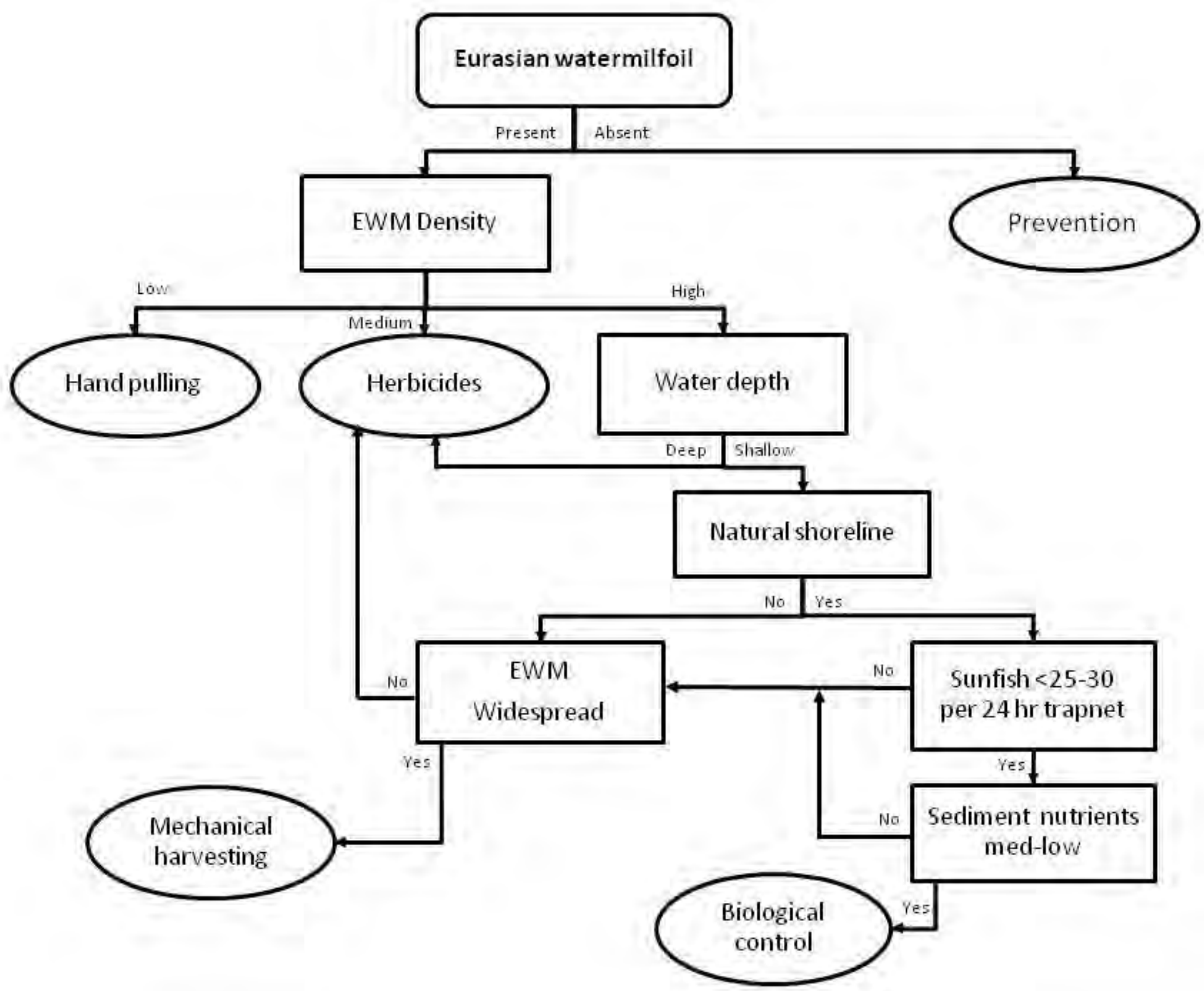


Figure 1. Decision Tree, taking shape under continued study of EWM biocontrol, illustrating how lake characteristics can help guide the decision to use biological control. (Trapnet benchmark dependant on equipment and methods specifications, based on Ward and Newman 2016.) Further studies are needed to refine and verify benchmarks, to guide management decisions with improved predictability of results.

4. Other control activities – Integrating biological control with other control methods is possible, but must be carefully planned. For details on research regarding integrated control, see Section I.

   a) Handpulling – If milfoil is in large dense beds, but not yet spread throughout the lake, biological control may work well in those dense beds, but milfoil may continue to spread while the weevils are getting established. Handpulling would be useful to control the isolated, individual plants and keep the milfoil confined to the beds where weevils have been stocked. Handpulling would not be recommended in the stocked beds.

   b) Chemical control – Herbicide treatments may be useful to provide control (but likely not eradication) of EWM under certain conditions. When the chemicals kill the milfoil plant, it also kills the juvenile weevils growing on it. Adults, however, may be able to evacuate to other surviving milfoil and create an increase in those locations. Therefore, targeted chemical “spot treatments” of milfoil may theoretically be compatible with biological control, however, spot treatments are not frequently recommended anymore in Wisconsin. If there is an isolated bay that would likely not be impacted by chemical treatments in the lake, this may be a location where biological control could be considered. The potential for dissipation and drift of the chemical should be carefully considered; even low, non-lethal concentrations of the chemical may be sufficient to cause deformities or growing irregularities in the plant, such as dark, tough stems, which may cause the plant to be unpalatable or unusable to the weevil, decreasing weevil success.

Deformed milfoil with fused leaflets and tough, opaque stems, typical of deformities caused by exposure to herbicides. Such stems are seen to be unused by weevils in captive rearing situations.

   c) Mechanical harvesting – Mechanical harvesting may be useful to provide control (but not eradication) of EWM under certain conditions. Harvesting of over 15% of the milfoil beds where biological control is being used is detrimental to weevil success. Therefore, harvesting should be excluded from, or minimized in, biocontrol areas.

5. Cost – There is currently no company offering direct stocking services. Weevils can be mass reared through a volunteer rearing program developed by Golden Sands Resource Conservation & Development Council, Inc. (See Section V for details.) Weevil starter stock must be either collected or purchased from (supplier

undetermined). With costs of weevil starter stock (at rates previously offered by EnviroScience, Inc. of Stowe, OH) and the equipment involved, costs are estimated approximately $0;.31 per weevil produced, assuming all volunteer labor.

Options for implementation

One option for implementing biological control is to foster naturally-occuring weevil population growth. If the lake had native milfoils, especially northern watermilfoil (*Myriophyllum sibiricum*) present prior to the introduction of Eurasian watermilfoil, then a native population of milfoil weevils may already be present. Weevil monitoring surveys (see Section IV) may determine the presence and current density of the weevil population.

Natural milfoil declines in association with the milfoil weevil have been recorded. (See Section I for discussion.) These natural declines often occur decades after Eurasian watermilfoil is introduced into the lake, but supporting weevil habitat (both in-lake and on-shore) may enable their populations to increase more rapidly. (See next section for details on this.)

A second option for biological control is stocking weevils to artificially boost the population, thus increasing the chances that a milfoil decline happens sooner. Section V includes instructions for mass rearing milfoil weevils for stocking. A stocking program would require a significant commitment from the lake group, including:

1. Protecting/restoring on-shore habitat to support weevil winter survival.
2. Protecting in-lake stocking locations to support weevil reproduction success.
3. Dedicating significant volunteer labor towards a successful mass rearing program.
4. Annual monitoring surveys to track both milfoil and weevil progress.

Managing your lake for biological control

Managing a lake for biological control should support both the in-lake and on-shore habitats of the milfoil weevil. In-lake protection will promote successful reproduction throughout the summer. On-shore protection will promote winter survival.

The milfoil beds where the weevil populations are most dense, or where they are stocked, should be protected from disturbance by mechanical harvesting, boat traffic, or chemical treatments. Weevils lay eggs on the apical meristem of the plant, and this is what boat traffic or mechanical harvesting may disturb or remove. Chemical treatments kill the entire plant the weevils live on, and thus cannot be used in the same location as weevils. (See preceding pages on “Evaluation Criteria” for further discussion on integrated control.) Awareness of weevil stocking locations should be discussed and promoted with all lake residents. Buoys and/or signage can be put in place throughout the boating season to inform boaters to avoid that location. (Permits required for placement of buoys/signage.)

On-shore habitat should be protected to increase winter survival of the weevils. This can be done by protecting or restoring natural shoreline habitat (unmowed plants, shrubs, and trees) within 35 feet of the water, minimizing maintained (mowed and raked) areas, and refraining from mowing or raking of maintained areas between Labor Day and Memorial Day.

Why wait until there is a problem?

Forward thinking lake groups can begin promoting on-shore habitat now, so their lake will be ready to support weevils, should they need them in the future. Healthy shoreline habitat will provide many other important benefits to the lake, as well, including wildlife habitat, erosion control, protection from pollution, and scenic beauty.

IV. MONITORING

Annual monitoring of both the milfoil and the weevil populations is important for tracking both populations and guiding management. With stocking, you should ideally see weevil population densities increase and relative abundance milfoil decrease. Weevil population densities of 1.0 weevils per stem should provide control in most lakes, but densities as low as 0.25 weevils per stem have even been recorded to provide control (Newman 2004). It seems different for each lake.

If weevil numbers do not increase from year to year, despite annual stocking, the stocking program may need to be increased, or there may be a limiting factor preventing weevil reproductive success or winter survival. Fish surveys to examine the bluegill population (per methods in Ward and Newman 2006) may provide helpful information about predation pressure. An evaluation of shoreline habitat should examine whether more undisturbed shoreline is needed to provide high and dry habitat with duff cover.

Milfoil declines may not be apparent to the casual observer. The sampling methods provided below will help to measure the subtle changes in the aquatic plant community from one year to the next. Remember, biological control will not eradicate milfoil, but rather reduce its density so native plants can rebound and provide a richer, more diverse aquatic ecosystem.

Methods for professionals

Monitor both the milfoil and milfoil weevil populations. You can survey for weevils at the same time as your aquatic macrophyte point intercept survey. Optimal times for weevil surveys are July through mid-August. Optimal times for macrophyte surveys are July through mid-August.

To survey the milfoil population, complete an aquatic plant community survey using Wisconsin's Standard Aquatic Plant Survey Method, available in full detail at [dnr.wi.gov](dnr.wi.gov).

Below is a quick summary of the monitoring protocol, with the steps for weevil surveys added in:

Field equipment needed:

- Boat (kayak, fishing boat, paddle boat, etc.)
- Personal Floatation Device (PFD)
- Point intercept survey grid (obtain standard grid from DNR)

Standard PI rake: a double-headed garden rake, constructed by cutting one handle off and fixing the two heads together. Rake on a rope used for deep sites. Photo: WDNR

- Long handled, double-headed rake with attached rope
- Pencil for marking datasheet
- Clip board or other hard surface for writing
- Point intercept datasheet
- Ziploc® bags
- Waterproof sharpie pen (to write on Ziploc® bags)
- Cooler to keep plants in
- GPS unit
- Polarized sunglasses (optional)
- Aqua-View Scope (optional)

A sample grid must be obtained from Wisconsin Department of Natural Resources. Navigate to the sample points in the field by GPS. At each sample point, drop a double-headed metal thatching rake straight down to the bottom, turned 360 degrees, then pulled straight back up. If the location is too deep to reach the bottom with the rake handle, a rake on a rope may be thrown, dragged 2.5 ft (0.75 m), then pulled straight back up,

The plants snared on the rake are identified by species and "rake fullness" for each species is ranked, 1 through 3. A rating of "1" indicates few plants present on the rake head, "2" indicates the rake head about ½ full and "3" indicates the rake is overflowing with no tines visible. If nothing is found, the entry is left blank. These rankings can later be plotted on the map and used to interpolate boundaries of milfoil beds of "sparse" ("1"), "dense" ("2") and "very dense" ("3") rankings. If a plant species is observed within 6.5 ft (2 m) of the boat but does not appear on the sample rake, it may be recorded as "v" for "visually observed".

Fullness Rating	Coverage	Description
1		Only few plants. There are not enough plants to entirely cover the length of the rake head in a single layer.
2		There are enough plants to cover the length of the rake head in a single layer, but not enough to fully cover the tines.
3		The rake is completely covered and tines are not visible.

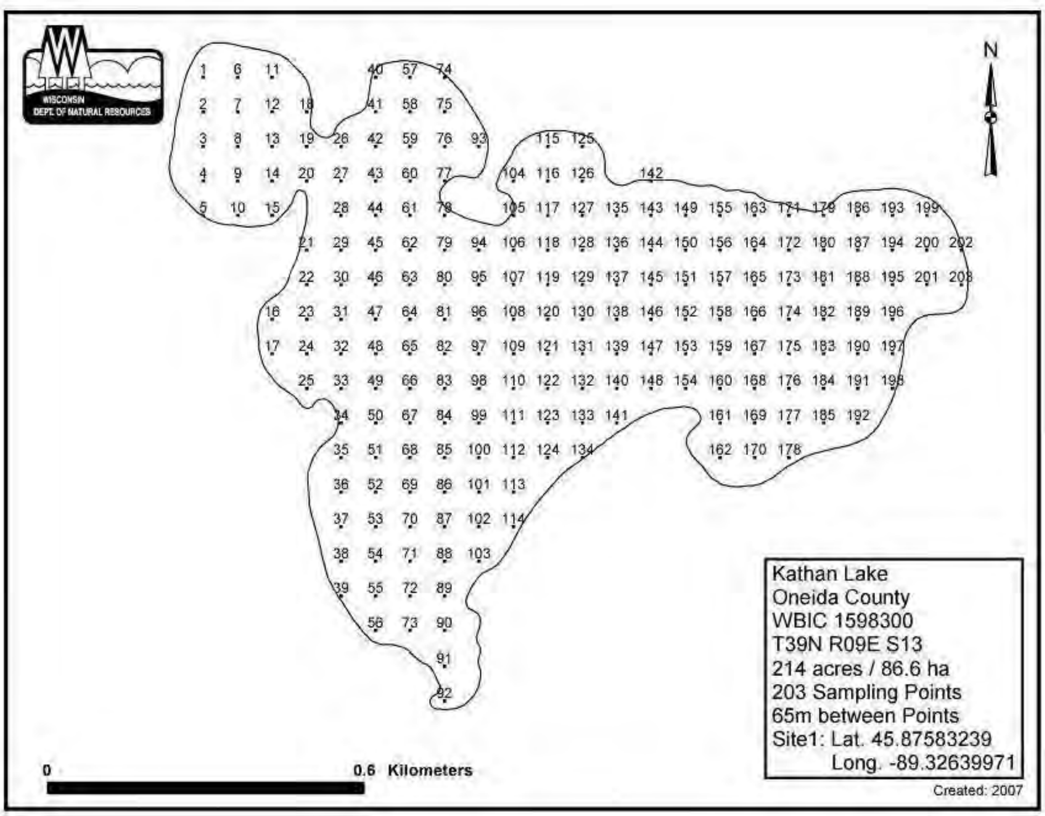
Illustration of rake fullness ranking, excerpted from Wisconsin's Standard Aquatic Plant Survey Method.

To sample for weevils, at each point where Eurasian watermilfoil is found, collect **two stems** to retain as weevil samples. Milfoil weevils live within the top 20 in (50 cm) of *M. spicatum* stems, therefore, retain the top 24 in of the stem as your sample. Collect stems by reaching into the water and grabbing the first stem your hand touches. Where *M. spicatum* is not close to the surface, use the PI rake to collect a fresh sample of stems. (Do not use the stems collected during your PI sample.) The first intact, 24 in-long stem to be randomly selected and untangled from the rake will be retained as the sample stem. Refrain from visually scanning the stems before picking them, which would introduce sampling bias. Be sure to collect any pieces of the stem that breaks off as you are collecting. Do not collect the lower portions of the stem below the top 25”. (These are unnecessary and may pollute the sample.)

Quickly place stem samples into a plastic bag. Excessive handling may cause adult weevils to drop off. Be sure to include any stem fragments that break off. Store and label stems from each sample point separately. No water needs to be added to the sample bag. Keep sample bags in coolers while in the field. In the lab, preserve each sample with 70% isopropyl alcohol or ethyl alcohol and keep refrigerated at 4°C until examination.

Voucher specimens of each plant species sampled during the aquatic plant survey should be collected in a labeled re-sealable bag, kept cool, and later pressed and mounted. For your PI data to be officially accepted by WDNR, the species identification should be declared and/or verified by an authorized botanist. Check with WDNR for authorized botanist in your area. Voucher samples should be retained in a climate-controlled herbarium.

Enter data into standardized Aquatic Plant Survey Data Workbook (Excel spreadsheet) available from WDNR. Spreadsheet will automatically calculate important metrics for tracking increases and decreases in the Eurasian watermilfoil population, such as relative frequency and average rake fullness ranking.



An example of a standard PI grid created by WDNR, as excerpted from Wisconsin’s Standard Aquatic Plant Survey Method. PI grids are created with standardized spacing to provide data with statistical confidence.

Determine weevil population density by examining weevils:

Laboratory equipment needed:

- Pans (9”x13” clear, glass pans)
- Light table
- Magnification (hand-held lenses, goggles, and 50x MagniScopes™)
- Lighting (bright lighting, overhead lamps)
- Black paper or plastic
- Tweezers
- Probes (long, finely-pointed)
- Eye droppers (plastic, cut end to a wide-mouthed)
- Clean water

Examine each stem under magnification by floating them in shallow water in a clear, glass pan over a light table. Using 10X magnifying goggles or a handlens, examine stems for eggs, larvae, pupae, and adults: Start at the meristem, inspecting between each of the leaves of the meristem for eggs and early instart larva. Next, work your way down the stem looking for larva, pupa, and adults. Also note any signs of weevil damage, included chewed meristems, larva pinholes or tunneling, and pupal chambers or blastholes created by pupa exiting. Record the number of weevils and presence or absence of weevil damage for each sample point.

Carefully extract all weevils of all life stages found in or on the stem. Use the aid of a 30x Carson Magniscope™ as needed to confirm species identification. Preserve specimens in a glass vial with 70% isopropanol, storing and labeling specimens from each sample point separately.

Larvae and pupae are easy to detect with the use of a light table. To detect eggs and young larvae, use the aid of overhead lighting and a black background under the meristem. They will almost appear to glow against the dark background. Do this by turning off the light table, turning on an overhead gooseneck lamp, and slipping a black sheet of paper under the glass pan.

Data interpretation:

A sample weevil datasheet is provided in Appendix B. Because samples are bagged and recorded by sample point, you will be able to map out where weevils are locally concentrated and evaluate what depth zones they are most abundant in in your lake. You may even be able to see a difference between high-traffic areas and quiet, undisturbed areas. Expect variation from bed to bed, and expect those patterns to change from one year to the next as the population moves around. Average natural population density in Wisconsin is approximately 0.65 weevils per stem (Jester et al. 2000). Population densities of 1.0 weevils per stem are likely to induce a milfoil crash (and thus we target that number when stocking), but densities as low as 0.25 weevils per stem have been documented to produce a milfoil crash (Newman 2004). After the milfoil population crashes, the weevil population should be expected to decline along with its food source (Newman and Biesboer 2000, WDNR 2014 unpublished report).

Plant survey data will track changes in the Eurasian watermilfoil population from year to year. Relative frequency of Eurasian watermilfoil may remain stable or even increase. A declining milfoil population is more accurately indicated by declining plant biomass (Newman and Biesboer 2000), meaning you should see EWM average rake fullness ranking decline. Perhaps more importantly, as you see Eurasian watermilfoil biomass decline, you should also see the average rake fullness ranking of native species increase, indicating your native plants are on the rebound.

Methods for volunteers

This volunteer monitoring method is part of the Citizen Lake Monitoring program. A complete volunteer monitoring manual is available through the CLMN program at:

http://www.uwsp.edu/cnr-ap/UWEXLakes/Pages/programs/clmn/AIS.aspx

The CLMN manual method includes protocols for monitoring both the milfoil population and the weevil population, to track whether they are increasing or decreasing. Optimal times for weevil surveys are July through mid-August. Please consult the manual before monitoring for complete details.

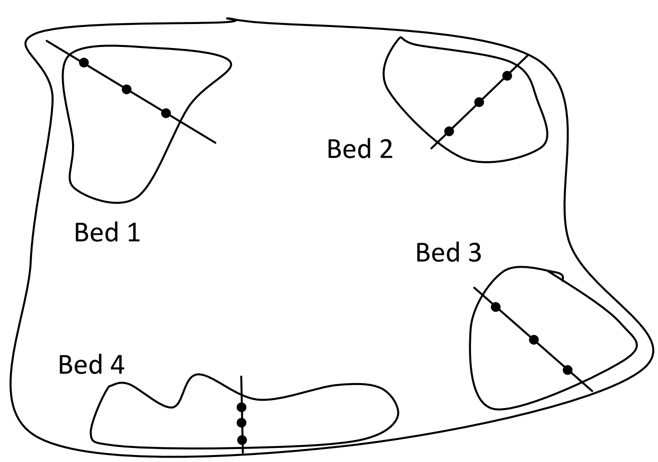
Below is a quick summary of the monitoring protocol:

Field equipment needed:

- Boat (canoe, kayak, fishing boat, paddle boat, etc.)
- Personal Floatation Device (PFD)
- Long handled rake with attached rope (thatching rake is ideal)
- Lake map for marking EWM beds and sample sites within the bed.
- Pencil for marking on map
- Clip board or other hard surface for writing
- Ziploc® bags
- Waterproof sharpie pen (to write on Ziploc® bags)
- Cooler to keep plants and weevils in
- GPS unit (optional)
- Polarized sunglasses (optional)
- Aqua-View Scope (optional)

Thatching rake. Good for cutting clean stem samples, rather than pulling plants by the roots. Photo: www.acehardware.com

Designate a minimum of four (10 max) representative milfoil beds in the lake, or within the area of the lake where biocontrol is being used. Label them Bed #1, #2, #3, and #4. Sample during peak season (July through mid-August.) Using a boat or canoe, boat in a straight line through the middle of the bed, from the deep side of the bed to the shallow side. This is your sample transect. Stop at three sample points along the transect to collect samples: a) shallow edge, b) middle, and c) deep edge.



At each sample point, collect 10 sample stems; if the milfoil is near the surface, reach down with your hand and randomly grab the first milfoil stem your hand touches. Avoid sample bias by looking away as you reach for the stem, so you will not be subconciously selecting stems with a certain appearance. Do not discriminate against raggedy or especially healthy-looking stems, however the stem must be rooted at the time of collection. Do not collect “floaters”. Collect the top 24” of the stem, careful to grab any fragments that break off. Discard the lower stem section beyond the top 24”. Repeat this 5 times on each side of the boat until you have randomly selected 10 stems total at each sample point. If the milfoil is deep, collect stems by throwing the sample rake on each side of the boat and carefully untangling and collecting 5 stems, for a total of 10 stems. Quickly place each sample stem into a bucket or ziplock bag with water, to preserve the sample. Keep sample stems from each sample point separate and labelled. Keep the specimens cool. You will examine them later in a laboratory setting.

AFTER you have collected your weevil stem samples, rate the milfoil density by extending the sample rake so that the rake head is roughly 3.0 feet from the gunwale of the boat, then lower the rake to the lake bottom. Pull the rake toward the boat until the head of the rake is still resting on the bottom and the handle is roughly perpendicular to the water’s surface. The rake head should be pulled approximately 2.5 feet along the substrate (bottom), then be pulled to the surface and into the boat. Once in the boat, rank the EWM stand density 1-3. One for only a few plants on the rake head; two for half full, the top of the rake can be easily seen; and three for overflowing where the rake head cannot be seen.

Collect additional data at each sample point, as you are equipped and able: Depth, substrate type, water temperature, water clarity, and/or dissolved oxygen. Because shoreland habitat is critical in winter survival of weevils, also record the amount of shoreland habitat available; only vegetated, unmowed areas with leaf litter should be considered habitat. Mowed lawns, rip-rap, and sandy beaches should not be considered shoreland habitat. Record where there is 0 ft, 1-10 ft, 10-20 ft, or 20+ ft of shoreland habitat.

Repeat these steps to sample all of your sample beds. Keep weevil stem samples refrigerated until examination.

Laboratory equipment needed:

- Pans (9”x13” clear, glass pans or large, white-bottomed pans)
- Light table (optional)
- Magnification (hand-held lenses, goggles, or 30x MagniScopes™)
- Lighting (bright lighting, overhead lamps)
- Tweezers
- Probes (long, finely-pointed)
- Eye droppers (wide-mouthed is best, a turkey baster will do in a pinch)
- Clean water

To examine stems samples, place each into a pan of water, one at a time. Using magnification and a probe, examine stem, starting at the top. Examine stems for eggs, larvae, pupae, and adults. Also note any signs of weevil damage, included chewed meristems, larvae pinholes or tunneling, and pupal chambers or blastholes. Record the number of each of these per 10 stems (from each sample point).

Consult the CLMN voluteer manual for detailed instructions and helpful diagrams and photos.

V. MILFOIL WEEVIL MASS REARING:

***A simplified outdoor rearing method for lake groups**

**The guidance of a qualified professional biologist during this process is recommended, in order to increase success rates.*

Mass rearing of weevils in predator-free enclosures may result in higher end-of-season populations than naturally-occurring populations or traditional, early-season stocking methods. Whether natural population densities reach levels capable of whole-lake control appears to vary from lake to lake, and estimates of densities required to affect control varies, from as little as 0.25 per stem to $\geq$1.0 per stem (Newman 2004). To increase densities to levels that may control Eurasian watermilfoil, whole-lake supplemental stocking has been attempted in numerous lakes throughout the U.S., with mixed results (Reeves et al. 2008). Present use of milfoil weevils to control Eurasian watermilfoil involves supplemental stocking of large numbers of weevils reared off-site to feed on and subsequently kill Eurasian watermilfoil. Tens of thousands [3000 per acre (Madsen et al. 2000)] of weevils are usually required to have an effect. These stocked weevils have historically been introduced to the lake early in the season, leaving the weevils to feed and multiply under natural lake conditions, including predation pressure.

Weevil population models have suggested that end of season populations are most critically linked to the survival of adult stage weevils; increasing a female adult's lifespan from five to 10 days can result in an 8-fold increase in end-of-summer population densities (Ward 2002, Newman et al. 2002). Therefore, mass rearing in predator-free enclosures, with reared weevils released to the lake later in the season, could maximize the number of weevils produced by the end of the season.

Prior to embarking on a milfoil weevil mass rearing program for your lake, read through <u>all</u> the instructions given here, and plan your project thoroughly with a qualified professional biologist. A "quick guide" is available in Appendix C to keep at your rearing station for handy reference.

Equipment

- 100-gal poly stock tanks, available from hardware stores like Fleet Farm, $75 ea
- Fiberglass screen, available in rolls from home improvement stores like Menards
- 1" binder clips or sturdy clothes pins
- 1"x2" lathing strips, cut to 4-foot lengths
- Staple gun
- Water supply – fresh, chlorine-free, insect-free
- Hose and sprayer nozzle
- ½" screen with a support frame
- Two saw horses
- Three 4' plastic wading pools (for sorting, bundling, and holding)
- 2 clean dish pans
- Fresh Eurasian water milfoil (EWM) to feed to weevils
- Starter stock of weevils (*Euhrychiopsis lecontei*), 70 per tank

Methods

1. **Advance preparations – Considerations, permits, equipment, and starter stock**
   Several months prior to rearing your weevils, discuss the project with your Department of Natural Resources (DNR) water resources management specialist. Consider specific attributes of your lake and whether milfoil weevils have good chance of success on your lake. Refer to Section III of this manual for evaluation criteria and discussion on using biocontrol as a management tool. Discuss whether your lake group has a good potential rearing locations and good potential release sites. You will also need about 6-12 dedicated volunteers. Be realistic about your lake group's capabilities.

   If your group chooses to rear weevils, it should be a 3-year commitment, minimum, to give the weevil population a chance to build. Monitoring should be implemented to track the population's progress. (See Section IV for monitoring methods.) Secure any permits needed for stocking milfoil weevils to your lake.

   Order the necessary weevil starter stock several months in advance. EnviroScience, Inc., of Stowe, OH, is no longer selling weevils in Wisconsin. You can now *order your weevils from Golden Sands Resource Conservation & Development Council, Inc., Stevens Point, WI (www.goldensandsrcd.org). Order 70 weevils for each rearing tank. Alternatively, you can collect your own in spring and raise starter stock in 10-gallon aquarium. (Not recommended. Methods under development.)

   Purchase all needed equipment in May, prior to receiving your starter stock in June.

2. **Set-up tanks**
   Select your rearing site: Select a secure, level site with good sun exposure. Healthy milfoil grows healthy weevils; the site will need full sun (at least 6 hours of sun per day). The site also needs to be fairly level. Secure the site to prevent tampering and so the tanks will not present a safety hazard to children.

Temporary fencing and informational signage will tell curious bystanders that the tanks are part of your special lake-improvement project.

   Chambers may alternatively be floated in the lake using foam noodles. If floated in the lake, fit drain holes with 500 micron mesh to let water in and keep insects and minnows out. (See Appendix D for diagram of in-lake tank design.)

   Purchase enough tanks to produce the number of weevils you want to stock to the lake. Each tank will produce about 670 weevils. Tanks are durable and can be used year after year. (Remove plugs during winter storage to reduce problems with leaking plugs.

 **Weevil availability is dependent on weather and climate conditions any given season.*

If you are a first-time rearer, it is suggested you start with four or five tanks the first year and see how it goes. Bump up production in year two if you feel ready.

Arrange the tanks in the rearing site the day before weevil starter stock arrives. You will also need to access each tank for feeding and maintenance, so leave room to maneuver and clip/unclip screens. Once filled with water, tanks will be too heavy to move.

Fill tanks the day before the weevil starter stock arrives to give the water time to equalize with the air temperature. Water should be free of chlorine, insects, or other pollutants. Well water is ideal and should require no treatment. Municipal tap water should be carbon filtered to remove chlorine. Lake water should be filtered through a 500 micron mesh to remove insects, algae, and debris.

Cover tanks with fiberglass porch screen immediately after tanks are filled to keep predator insects and debris out. Cut screen large enough to fully cover tanks and wrap over the sides. Staple a 1”x2” lathing strip down the length for support. Secure to chamber on all sides with 1” binder clips or sturdy clothes pins, ensuring there are no gaps where predator insects could crawl in or your weevils could escape.

Set up your sorting, cleaning, and bundling stations: Set your pools for sorting, bundling, and holding in a level spot, fill with clean water, and cover with screens. Set up your cleaning station, with hose, sprayer nozzle, and 1/2” hardware cloth.

Carbon filtration tank for treating municipal tap water is visible in the upper right. You may be able to rent a carbon filtration tank from your local water treatment company.

Above: You will need to tend your tanks throughout rearing season. Remember to leave yourself room to access each tank and remove/re-secure screens.

Below: Building a frame for your hardware cloth and supporting it at waiste height will make cleaning more comfortable work.

3. **Collect food for weevils**
   Use clean hands/arms to handle milfoil throughout this process. **No sun lotion, bug spray, or other contaminants.**

   Using a rake on a rope, collect Eurasian watermilfoil stems from the deep areas of the lake, where algae, insects and debris on the plants will be minimal. Collect only Eurasian watermilfoil (*Myriophyllum spicatum*), no other milfoil species. (See Appendix E for a guide to discriminating between milfoil species.) Select the most beautiful, healthy milfoil you can find.

   Discard dirty, unwanted roots. Untangle the milfoil from the rake carefully to avoid breakage as much as possible.

   Place milfoil into a bucket, cooler, or bin, and keep cool and wet while transporting it to your rearing site. Do not allow milfoil to dry out.

   Place milfoil into the sorting pool and submerse in clean water. Do not allow milfoil to dry out. Keep pool covered with screen until ready to sort, in order to keep out predator insects.

Avoid contamination: No sun lotions or bug sprays!

A thatching rake will collect cleaner milfoil. It cuts stems, rather than pulling it by the roots. Photo: www.amleo.com

4. **Sort EWM stems**
   Sort through EWM stems to select healthy-looking stems with lush, bushy meristems (growing tips). Weevils are very picky! They only lay their eggs on the meristems, and prefer bushy ones. Broken tips provide no place for weevils to lay eggs.

   Break off the top 25” of the stem. This is your **food stem**. Keep food stems wet as you are sorting. Do not let them dry out.

   Discard the lower portions of the stems, as well as any **unhealthy-looking, blackened, or skinny stems**, stems with flowering tips, or broken tips. Also discard any stems of the wrong species. Only *M. spicatum* should be used. Discarded stems are nutrient-rich and make great compost.

A beautiful meristem! This female weevil is laying multiple eggs on this bushy meristem. Keep your weevils happy by giving them the best quality EWM stems possible.

Healthy milfoil grows healthy weevils!

Many hands make light work…. Chatting with friends around the milfoil sorting pool can make the work enjoyable!

Using 25" board as your measuring device can be a time saver for trimming your food stems.

5. **Clean food stems**
   Lay food stems on a ½" hardware cloth or screen in a <u>single</u> layer. Spray with clean water. The spray should be hard enough to blast debris off of the stems, but not hard enough to break or damage the stems.

   Transfer clean food stems to the bundling pool and submerse in clean water. Keep pool covered with screen until ready to bundle.

Lay stems in a single layer, so the spray will effectively clean the food stems.

6. **Bundle food stems**
   Use clean hands/arms to handle EWM. No sun lotion, no bug spray.

   To make a food bundle, organize 15 food stems together, holding stems by the bottom ends, with all the tips hanging downward. Make sure all the bottoms are even with each other, and secure a rock to the base of the stems with a rubber band.

A food bundle being held upside down. All stems line up with the base tied to the rock and the bushy meristems (growing tips) free to float to the water surface.

7. **Place food bundles into holding pool**
   As food bundles are made, they can be stored in the holding pool until there are enough to start stocking the tanks. Keep them submerged in clean water. Keep the pool covered with a screen.

8. **Place food bundles into tanks**
   Use clean hands/arms to reach into the tanks. No sun lotion, no bug spray. Keep tanks covered with screen between any maintenance activities.

   Place 7 food bundles into each tank. Keep track as you work, so all tanks get the correct amount of food. If you trimmed the EWM to the right length, meristems should be near the water surface. If any are too long and trailing along the water surface, trim the bottom to correct their length.

Freshly cleaned bundles in a tank. Once you know you have 7 bundles in each tank, push bundles together, so larvae can crawl from stem to stem.

   Keep bundles closely touching each other (but not tightly packed). Weevil larvae cannot swim and will need to crawl from stem to stem. Push stems to the north side of the tank. Stems next to the south side of the tank would be in the shade, and would turn brown quickly.

9. **Inoculate each tank with 70 weevils (starter stock)**
   Keep tanks covered with screen in between all maintenance activities. Use clean hands/arms to handle EWM and equipment. No sun lotion, no bug spray.

   Your starter stock will arrive in a cooler, on ice. Get them into the tanks as soon as possible. As soon as they warm up they will need to eat, so keep them chilled until you can inoculate them. However, the longer they remain on ice or refrigerated, the lower their survival rate.

   Each bag of weevils received from Golden Sands RC&D should have the number of weevils enclosed written on the bag. The weevils will be on short EWM stems (fragments) that are twist-tied together. Eggs and larvae are tiny and hard to see, but they are there.

Weevil starter stock arrives in sealed bags, on ice. The sooner you can get them into their tanks, the better. The longer you keep them iced/refrigerated, the more of them may die.

Inoculate each tank with approximately 70 weevils:

a) Determine the number of weevils going into each tank: Divide the total number of weevils shipped to you by the number of tanks. Record this number. It should be about 70.

b) Open bags, remove the twist-ties from the bundled EWM fragments (short stems) and lay the loose fragments in a large dish pan of clean water.

c) Place small containers of clean water next to the dish pan, one container representing each tank. (e.g. If you have 4 tanks, set out 4 containers.)

d) Grasping fragments by the base (never the tips – that is where the eggs are!), randomly select one EWM fragment from the dish pan and lay it into a container. Repeat, placing one fragment into each container in turn. This randomized process ensures that each container has an equal chance of receiving an equal number of weevils.

e) Continue randomly selecting fragments and placing them into each container in turn until no fragments remain in the dish pan. Lay them in with all tips pointed one way, bottoms facing the other. Each container now contains approximately 70 weevils to go into a tank.

f) Inoculate each tank with apx 70 weevils by distributing the fragments from on container into one tank:
    a. For each container, bundle together half of the fragments and twist-tie the bottoms of the stems together. Grasp fragments by the bottom end, never the tips. Bundle the other half also. You now have two bundles to go into a tank (apx. 35 weevils on each bundle).
    b. Fasten these two fragment bundles to two food bundles in a tank. Fasten them near the tips, so the tips of the fragments are next to the tips of the food bundles.

g) Repeat for each tank.

10. Feed weevils every 21 days (or 17 days in very sunny sites)

21 days (or 17 days) after inoculation: Repeat the food collection, cleaning, and bundling process. Add **11** food bundles to each tank. (Leave old bundles in.)

42 days (or 34 days) after inoculation: Repeat the food collection, cleaning, and bundling process. Add **15** food bundles to each tank. (Leave old bundles in.)

Make sure new bundles are evenly dispersed around the old bundles, and closely touching the old bundles. Keep screens secured on tanks. Tighten as needed. Top off tanks with clean, chlorine-free water as water gets low, using caution not to overly-disturb food stems.

Monitor food quality; as weevils feed, stems will become hollowed or blackened in sections, especially at the tips. This feeding damage is a good sign, but If it appears the weevils are running out of healthy, green, undamaged stems, you may have to bump up your feeding day a few days earlier. As you add fresh food bundles, you may notice your original food bundles deteriorating; stems turn black from the base up, lower leaves browning. This is normal, and is the reason we regularly add new food stems. However, if bundles deteriorate prematurely, or become questionable in any way, add fresh food as soon as possible. Do not remove old, browned bundles, as it will take time for weevils to migrate onto the fresh food.

GOOD: *The food bundle held here has browning leaves at the base and white adventitious roots sprouting. This is normal, and this bundle is still in good shape, with lots of green leaves and plump, bright-colored stems.*

Bad: *This food bundle has brown, slimy leaves except at the very tips, and many stems are limp and brown as well. The milfoil may have been old when bundled, or is not getting enough sunlight. Regardless, fresh food must be added soon, or the weevils will starve!*

NORMAL: *July, half way through the rearing season, and one more feeding to go. This tank is still in good shape, with plenty of green, healthy food stems left. (White object is thermometer.)*

NORMAL: *August, release day is here, and this tank is still in good shape; old food stems have browned and deteriorated, and broken bits have floated away due to feeding damage, but the fresh food stems are still green. The water has become murky, but that does not seem to bother the weevils. A little filamentous (stringy) algae does not seem to bother them either.*

Monitor temperatures near the stem tips (where the weevil eggs are). Weevils develop slowly in cold water, but at expected temperatures (25C/77F) they can develop from egg to adult in just 21 days. At optimal temperatures (29C/84F) for weevil development, they can complete the life cycle in a speedy 17 days. If your site is very sunny and your temperatures are running near optimum, feed your weevils every 17 days.

At 31C/87 F, weevil development rate declines, and sustained temps of 34C/93.2F are **lethal**. If tank temperatures approach 86 F, top off tanks with fresh, cool water. You can also suspend shade cloth above the tanks to create shade. (Do not lay shade cloth directly on top of tanks.) This should lower the tank temperatures by approximately 4 degrees (F).

Shade cloth can drop tank temperatures 4 degrees F. Suspend shade cloth above the tanks to allow for air flow.

54 days (or 44 days in very sunny sites) after inoculation: Release your weevils using the process below.

Release early if you see your freshest food stems being rapidly eaten (turning black, breaking into bits easily); your weevils are running out of food!

11. Release weevils to lake

Select your release site well in advance and leave it undisturbed. Do not collect food stems from that site. Select a quiet, relatively undisturbed bay with EWM beds that are just reaching the surface. Do not use sites where EWM is already trailing along the surface.

7 to 10 days after final feeding, release your weevils:

(See photographic steps on next page.)

a) Gently scoop all bundles out of tanks and lay them a cooler with water from the tank. Leave rocks attached. Lay food bundles into coolers in an organized fashion, all meristems on one end, rocks on the other.
b) Keep bundles submerged in tank water and keep cool throughout the release process.
c) Use a pool net or colander to sift broken plant fragments out of the tank, and add those to the plants in the cooler; adult weevils left behind in the tanks may be clinging to these plant fragments.

Normal: *When scooping bundles out of tanks, you might find the twist-ties (green object) from your original inoculation bundles. These stem fragments and food stems will be browned and quite deteriorated, due to both age and heavy weevil feeding damage.*

Release Day: Gently scoop bundles out of tanks.

Gently lay bundles into a cooler with tank water in it. Lay them in an organized fashion, with minimal tangling.

d) Using non-motorized watercraft, transport coolers out to the release site (EWM bed). Take care minimize disturbance in the release site. The best place to put your weevils is where EWM is just reaching the lake surface. (Very shallow areas where EWM has been trailing along the water surface all summer get hot and stagnant. Yuck!)

e) Gently lift one bundle at a time out of the cooler and nestle each down into thick EWM:
   i. Untangle stems carefully!
   ii. Leave rocks attached when possible, to anchor the bundle in place.
   iii. **Handle bundles gently** to minimize stem breakage. Weevil-damaged stems are fragile and larvae may fall out of broken stems.

12. **DONE!**

13. **Expected average return rate:** 9.6
Starting with 70 weevils in a tank, approximately 672 weevils should be produced
Example scenario: 4 tanks, 280 weevils and 1,960 EWM stems IN ➔ 2,688 weevils OUT

To determine actual rate of return, collect a 10% subsample from at least four tanks, and examine these for total weevils (all life stages). Each subsample takes approximately 8 hours to examine. This step is best done by a qualified professional biologist.

14. **Estimated time investment:** 0.05 hours per weevil reared, or 22 weevils per hour invested.

15. **Timeline:** Total rearing time 54 days (or 44 days in very sunny sites)

January – Planning. Order weevil starter stock. Apply for any needed permits.

May – Purchase equipment. Confirm volunteer crew members.

June – (mid-June) Set-up tanks with food 1-2 days prior to arrival of weevil starter stock. Inoculate tanks with starter stock.

July – Feed weevils two more times, every 21 days (or 17 days).

August – (apx 1st week of Aug) Release weevils 54 days (or 44 days) after inoculation.

For additional information:

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Milfoil weevil: *Euhrychiopsis lecontei*

Microimaging by Jeffery Dimmick, Wisconsin Department of Natural Resources

Above: Top and front view of two *E. lecontei*. Note distinctive striping, and raised bumps at the top of the 7th ridge on the elytra (wing coverings).

Bottom left: Another top view showing raised bumps.

Bottom right: Close-up of legs. Note long setae (leg hairs).

Phytobius weevil: *Phytobius leucogaster* (a.k.a. *Litodactylus leucogaster* Marsham)
Microimaging by Jeffery Dimmick, Wisconsin Department of Natural Resources

Above: Top view of two *P. leucogaster*. Note lack of striping on abdomen. Instead, there is a light-colored patch in the center of the upper abdomen.
Bottom left: Front view. Note the raised bumps at the top of the 5th ridge on the elytra (wing coverings).
Bottom right: Close-up of legs. Note lack of long setae (leg hairs).

Bagous weevil: *Bagous restrictus*

Photo: www.plants.ifas.ufl.edu, Copyright: 1997 USDA-ARS

Photo: www.bugguide.net, Copyright: Glenn A. Salsbury

Top photo: Back has a speckled appearance, rather than stripes.
Bottom photo: Prosternum (thoracic segment) reaches forward to surround the head, especially on the belly side, giving the appearance of the bug wearing a high turtleneck sweater. Bagous does not have a raised bump on the elytra, or long setae on their legs.

APPENDIX B: SAMPLE WEEVIL SURVEY DATA SHEET

Waterbody:
Sample Date:
Lab Date:

Ave. # weevils per stem:

				Stem condition				Weevil Damage? (Yes=1)				Weevils present				
Lab Date	Bed #	Point #	Stem #	Length (inches)	Algae/Marl Covered (1=yes, 0=no)	# Broken Tips	# Apical Tips (branches)	larval pinholes present	larval tunnels present	used pupal chambers present	dmg meris. present	# Eggs	# Larvae	# Pupae	# Adults	Comments
Totals =																
Averages per stem =																

* - 1 = present, 0 = not present

Total weevils (all life stages) =

% Of Stems With Weevil Damage (#/total stems) =

Ave Weevils Per Stem =

Survey Notes:

APPENDIX C: WEEVIL REARING QUICK GUIDE

Keep this quick guide handy at your rearing station. For details, refer back to Section V.

Use clean hands/arms to handle milfoil throughout this process. **No sun lotion, bug spray, or other contaminants.**

1. **Advance preparations**
   Discuss and plan with DNR. Apply for permits. Order weevil starter stock. Purchase equipment.

2. **Set-up tanks**
   Select sunny level site with access to clean, chlorine-free water. Arrange and fill tanks 1 day prior to receiving weevil starter stock. Cover tanks with screen. Set up sorting, cleaning, and bundling stations. Cover pools with screen.

3. **Collect food for weevils**
   Collect Eurasian watermilfoil (*Myriophyllum spicatum*) (EWM) stems from deep areas of the lake where plants are cleanest. Collect no roots. Keep stems cool and wet. Place milfoil into sorting pool and submerse in clean water. Do not allow milfoil to dry out. Keep pool covered.

- ✓ Healthy milfoil
- ✓ 25" long
- ✓ Busy meristems

4. **Sort milfoil stems**
   Sort through milfoil stems. Discard any plants that are not *M. spicatum*. Select only healthy *M. spicatum* stems with bushy meristems. Trim to 25". Keep wet. Discard any **unhealthy-looking, blackened, or skinny stems**, stems with flowering tips, or broken tips. Do not allow milfoil to dry out.

5. **Clean food stems**
   Lay food stems on a ½" hardware cloth in a <u>single</u> layer. Spray with clean water. Spray hard enough to blast debris off of stems, but not hard enough to break or damage stems. Place clean food stems into bundling pool. Keep wet. Keep pool covered.

6. **Bundle food stems**
   Bundle 15 food stems together, all bottoms even with each other and tips hanging down. Secure a rock to base of stems with rubber band. Place food bundles into holding pool. Keep wet. Keep pool covered with screen.

7. **Place food bundles in tanks**
   Place exactly **7 food bundles** into each tank. Bundles should be closely touching each other. Weevil larvae cannot swim and will need to crawl from stem to stem.

8. **Inoculate each tank with 70 weevils (starter stock)**
   Inoculate the weevils as soon as the shipment arrives. Follow inoculation procedure in the detailed instruction manual. Inoculate each tank with approximately **70 weevils**.

9. **Feed weevils every 21 days (or 17 days in very sunny sites)**
   - Feeding #2 = 21 days (or 17 days) after inoculation. Add **11 food bundles** to each tank.
   - Feeding #3 = 42 days (or 34 days) after inoculation. Add **15 food bundles** to each tank.

   Evenly disperse new bundles around old bundles. Bundles should be closely touching each other. Keep screens secured on tanks. Top off tanks with clean, chlorine-free water as water gets low, using caution not to overly-disturb food stems.

   Monitor temperatures near the stem tips (where the eggs are). Expected tank temperatures = 25C (77F). Optimal temperatures = 29C (84F). Sustained temps of 34C (93.2F) is **lethal**. If tank temperatures approach 86 F, top off tanks with fresh, cool water, and suspend shade cloth above tanks.

   Release early if you see your freshest food stems being rapidly eaten (turning black, breaking into bits easily); your weevils are running out of food!

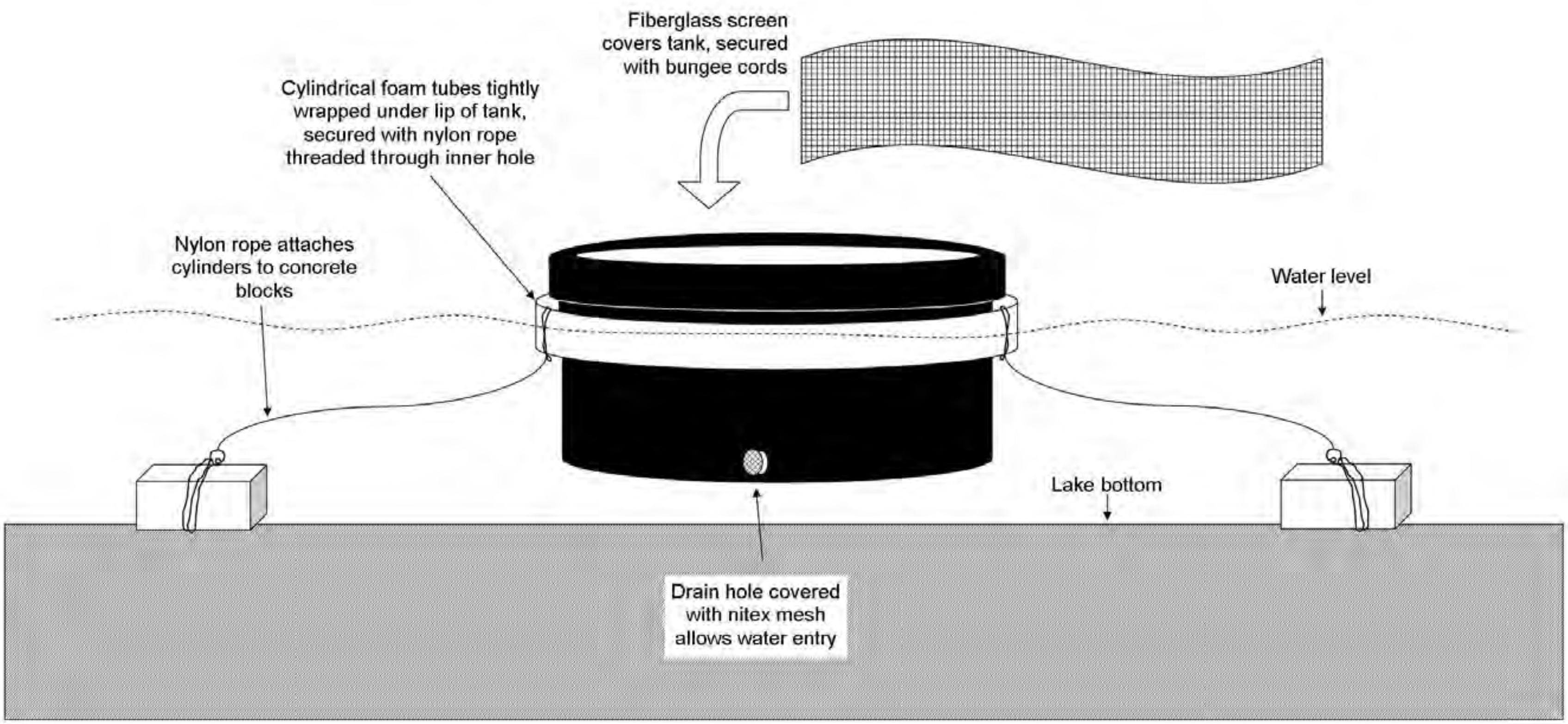
10. **Release weevils to lake = 7-10 days after feeding #3**
   Select a quiet, relatively undisturbed bay with EWM beds that are just reaching the surface. Do not use sites where EWM is already trailing along the surface.

   Gently scoop all bundles out of tanks and lay them a cooler with water from the tank. Leave rocks attached when possible. Organize bundles with all meristems on one end, rocks on the other. Keep bundles wet and cool.

   Using non-motorized watercraft, transport coolers out to the release site (EWM bed). Take care to minimize disturbance in the release site. Gently lift one bundle at a time out of cooler and nestle each down into thick EWM.

For additional information: Amy Thorstenson, Golden Sands Resource Conservation & Development Council, Inc., (715) 343-6215, Amy.Thorstenson@goldensandsrcd.org

APPENDIX D: DIAGRAM OF FLOATING WEEVIL CHAMBER



APPENDIX E: GUIDE TO DISCRIMINATING BETWEEN MILFOIL SPECIES

Milfoil weevils (*Euhrychiopsis lecontei*) grow bigger and faster when feeding on Eurasian watermilfoil, as opposed to any other milfoil. When rearing weevils, always use Eurasian watermilfoil. This guide will help you discriminate between the exotic EWM and other native species. (Identification keys referenced from *Aquatic Plants of the Upper Midwest*, by Paul Skawinski.)

Eurasian watermilfoil (*Myriophyllum spicatum*)

Stems: Thin, flexible, hang limp like spaghetti when out of the water. Usually pale to pinkish red. Often somewhat transparent with pink vein visible in center.

Leaves: Arranged in whorls (usually 4), evenly spaced up the stem about 2-3 cm apart. Leaves have 12-20 pairs of leaflets.

Non-native, invasive.

May hybridize with *M. sibiricum*.

Photos (above and below): Paul Skawinski, *Aquatic Plants of the Upper Midwest*

Eurasian vs Northern

EWM (left): Leaves have the appearance of feathers, with many fine leaflets (>12 pairs per leaf).

NWM (right): Leaves have coarser appearance and texture, capable of holding their shape out of water. Only 4-12 pairs of leaflets per leaf. Leaflets long and arching towards leaf tip, giving the appearance of a candelabra.

Northern watermilfoil (*Myriophyllum sibiricum*)

Stem: Stout, usually thick and does not hang limp like spaghetti when out of water. Usually whitish or tan, and not transparent, like *M. spicatum*.

Leaves: Arranged in whorls (usually 4), evenly spaced on the stem. Leaves are somewhat stiff and coarse, holding their shape when out of the water. Leaves have 4-12 pairs of leaflets.

Native.

May hybridize with *M. spicatum*.

Paul Skawinski, *Aquatic Plants of the Upper Midwest*

Farwell's milfoil (*Myriophyllum farwellii*)

Paul Skawinski, *Aquatic Plants of the Upper Midwest*

Stems: Delicate, collapsing when out of the water.

Leaves: Some leaves arranged in whorls on the stem, some alternate or scattered on stem. Leaves have less than 14 pairs of leaflets. Like the stems, leaves are delicate and collapse when out of the water.

Native.

Various-leaved milfoil (*Myriophyllum heterophyllum*)

Paul Skawinski, *Aquatic Plants of the Upper Midwest*

Stems: Stout, usually does not hang limp when out of water. Green-brown in color.

Leaves: Leaves arranged in whorls of 4-6 on the stem, some leaves alternate or scattered on stem. Whorls closely spaced on stem, less than 1.5 cm apart. Leaves have 7-14 pairs of leaflets. Plant appears very bushy compared to other milfoils.

Native.

APPENDIX F: WEEVIL OBSERVATION RECORDS WORKSHEET

Weevil Mass Rearing
Pilot Study

OBSERVATION RECORDS

Date	Time In	Time Out	Observer	Tank Temperature*	Shade cloth is on?	Topped off waterlevels	Checked Screens	Notes and Observations

* 84 F = Optimal, 87 F = Max, 93 F = Lethal. Add fresh water to any tanks 86 F or above.

GOLDEN SANDS
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Conservation That Works!

Hatch Lake, Waupaca County
Milfoil Weevil Survey
August 6-7, 2020

Golden Sands Resource Conservation & Development Council, Inc (RC&D) completed lab work for milfoil weevil samples collected by Onterra LLC staff during a Point Intercept Aquatic Plant Survey (PI Survey) on Hatch Lake on Aug. 6-7, 2020. The samples were collected to provide data about milfoil weevils that may be useful in lake management planning. The native milfoil weevil, *Euhrychiopsis lecontei*, is naturally present in many lakes in Wisconsin, and when present in dense populations, may be a biological control agent on Eurasian watermilfoil, *Myriophyllum spicatum* (EWM). For lakes with low weevil populations, adjustments in management may support weevil populations and allow biocontrol to become a growing piece of the larger management plan.

About milfoil weevils

The native milfoil weevil, *Euhrychiopsis lecontei*, is commonly found in Wisconsin lakes, and normally feeds on northern watermilfoil (Myriophyllum sibiricum), a native milfoil species. This fully aquatic weevil lays their eggs on the apical meristems (leaf buds), and the larvae hatch and tunnel through the stem, then exit and re-enter the stem to mine some more. About 15-20" down the stem, they burrow in and pupate, later emerging as an adult. Weevil lay eggs continuously throughout the summer.

In the fall, the adult weevils fly to shore and hibernate under the leaf litter. After ice-out, the weevils fly back to the lake and start the cycle over again.

About biocontrol

When the non-native Eurasian watermiolfoil moves into a lake, the weevils preferentially feed on the EWM. Larval feeding mines out the plant's vascular tissues and opens the plant up to bacterial growth. Sufficient damage reduces plant health and vigor and may even cause the plant to die.

Artificial stocking programs target population densities of about 1.0 weevil/stem (average), but natural EWM declines have been documented at as low as 0.25 weevils/stem. Every lake is different and various lake conditions may suppress a weevil population: Stunted panfish population (sunfish family), disturbance (boat traffic, mechanical harvesters), insufficient shoreline habitat, sparse or deep EWM populations.

Lakes that are optimal candidates have: Dense EWM populations that are close to shore and in shallow water (plants near water surface); EWM that has expanded to its fullest capacity; a balanced fish community, lots of natural shoreline.

When biocontrol successful, EWM is not completely eliminated, but rather it becomes a normal part of the plant community. Plants are shorter, not reaching the surface and shading out the other species. Plants are less dense and not creating monotypic beds, but rather sparse and mixed in amongst native species.

As with any predator-prey relationship, EWM and weevil populations will both fluctuate from year to year. EWM may increase and rebound, and in time the weevils increase to catch up.

Methods

Onterra LLc staff collected EWM stem samples during the point intercept (PI) survey of the aquatic plant community. At each survey point where EWM was found, 2 sample stems were collected by reaching into the water or using the sample rake. Only the top 24 inches of the plant was retained. Samples from each survey point were bagged, labelled, and preserved with ethanol.

Samples were later examined by Amy Thorstenson of Golden Sands RC&D by floating sample stems in a clear glass pan over a light table. Using 10x magnifying goggles, stems were inspected for weevils (eggs, larvae, pupae, adults) and weevil damage. Any weevils found were collected as voucher specimens, preserved in labelled sample vials.

Results

- 35 stems from 18 sample points
- 3 weevils found (2 larvae, 1 adult), all from 1 sample point
- 3 sample points had feeding damage indicative of *E. lecontei*
- Presence of *E. lecontei* = CONFIRMED
- Abundance of *E. lecontei* = Lakewide average of **0.09 weevils/stem**
- Link to lab data

Discussion

Weevils are present but in low population densities. The 3 weevils found all came from sample point 34. Two other points had weevil damage indicating the presence of weevils: pt 269 and pt 137.

Reviewing the PI survey map to look at spatial distribution of weevil presence may be helpful for monitoring populations from year to year. While Hatch Lake does not currently have weevil densities sufficient for controlling EWM, residents can build into their lake management plans elements that support weevil populations and allow them become an increasing part of their milfoil management program.

Shoreline habitat: Residents can take immediate action to support weevil populations by reducing mowing within 35 feet of the lakeshore. Where they do need to mow near shore, stop mowing/raking on Labor Day and wait until Memorial Day to resume mowing and leaf cleanup. Just 1 cm depth of leaf litter is enough to support weevil hibernation. Weevils move around the lake a lot from year to year, so the more natural shoreline available the better. (Bogs, wetlands, saturated soils are too wet for weevils.)

Boat traffic: Some EWM beds may be prime weevil habitat where populations may be able to expand. Looking at the current spatial distribution of the population may help identify EWM beds where disturbance should be avoided. Some lake groups have buoyed EWM beds to exclude boat/harvester traffic, allowing weevils to expand.

Fish predation: DNR may have recent data on the fishery that shows whether the panfish population is stunted or not. Practicing catch and release of the larger predator fish may help maintain a balanced fishery.

EWM management programs: Management programs may be designed to work in tandem with biocontrol, depending on the unique situation of each lake. Continued monitoring for weevils can help provide information for designing short and long term strategies.

Waterbody: **Hatch Lk, Waupaca Co**

Sample Date: 8/6-8/7/2020

Ave. # weevils per stem: **0.09**

Lab Date	Bed No.	Point No.	Stem No.	Length (in)	Algae/Marl Covered (1=yes, 0=no)	# Broken Tips	# Apical Tips	Weevil Damage? (1=yes/0=no) dmg meris. present	pinholes present	tunnels present	pupation chambers present	# Eggs	# Larvae	# Pupae	# Adults	Comments
10/28/20		290	1	*	0	*	*	0	0	0	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
10/28/20		201	1	*	0	*	*	0	0	0	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
11/3/20		136	1	*	0	*	*	0	0	0	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
11/3/20		269	1	*	0	*	*	0	1	0	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
11/3/20		271	1	*	0	*	*	0	0	0	0	0	0	0	0	Only 2 small apical tips in the bag. Chew marks, poss. from adult weevils?
			2	*	0	*	*	0	0	0	0	0	0	0	0	
11/3/20		267	1	*	0	*	*	0	0	0	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
11/3/20		59	1	*	0	*	2	0	0	0	0	0	0	0	0	Red worm tunnels
						*										
11/3/20		268	1	*	0	*	*	0	0	0	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
10/27/20		23	1	*	0	*	*	0	0	0	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
10/27/20		224	1	*	0	*	*	0	0	0	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
10/27/20		180	1	*	0	*	*	0	0	0	0	0	0	0	0	Feeding damage (from acentria?)
			2	*	0	*	*	0	0	0	0	0	0	0	0	
10/27/20		179	1	*	0	*	*	0	0	0	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
10/27/20		58	1	*	0	*	*	0	0	0	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
10/27/20		137	1	*	0	*	*	0	0	1	0	0	0	0	0	
			2	*	0	*	*	0	0	0	0	0	0	0	0	
10/27/20		243	1	25	0	1	1	0	0	0	0	0	0	0	0	Healthy, beautiful stems, no branching
			2	25	0	1	1	0	0	0	0	0	0	0	0	
10/27/20		56	1	*	0	1	*	0	0	0	?	0	0	0	0	External chewing on stem: from adult weevil? Or odonate? Break in stem shows tunneling, poss. from weevil?
			2	*	0	*	*	0	0	0	0	0	0	0	0	
10/27/20		159*	1	*	0	*	*	0	0	0	0	0	0	0	0	My datasheet has eraser mark: could be 189?
			2	*	0	*	*	0	0	0	0	0	0	0	0	
10/27/20		34	1	*	0	0	2	0	0	0	0	0	0	0	0	

Waterbody:	**Hatch Lk, Waupaca Co**															
Sample Date:	8/6-8/7/2020											Ave. # weevils per stem:			**0.09**	
								Weevil Damage? (1=yes/0=no)								
Lab Date	Bed No.	Point No.	Stem No.	Length (in)	Algae/Marl Covered (1=yes, 0=no)	# Broken Tips	# Apical Tips	dmg meris. present	pinholes present	tunnels present	pupation chambers present	# Eggs	# Larvae	# Pupae	# Adults	Comments
			2	*	0	0	2	1	1	1	1	0	2	0	1	Adult = E. lecontei (7th eletryal ridge raised, long setae)
		Totals =	35	*	0	*	*	1	2	2	1	0	2	0	1	
Averages per stem =					0%			6%	6%	6%	3%	0.00	0.06	0.00	0.03	
												Total weevils (all life stages) =			3	
					% Of Points With Weevil Damage (#/total stems) =				**9%**			Ave Weevils Per Stem (3/35) =			**0.09**	

Survey Notes: CONFIRMED, PRESENCE OF EUHRYCHIOPSIS LECONTEI - A. Thorstenson
* = broken stems, unable to count. * entered is omitted by a "COUNT" formula.
Survey method = PI
Samples were collected by Onterra team during PI survey, using collection methods specified by ALT. Most samples very brittle, stems snap appart w/ little pressure, leaves snap off w/ little pressure. Samples preserved in ethanol, unknown % concentration = sample stem friability likely related to this.