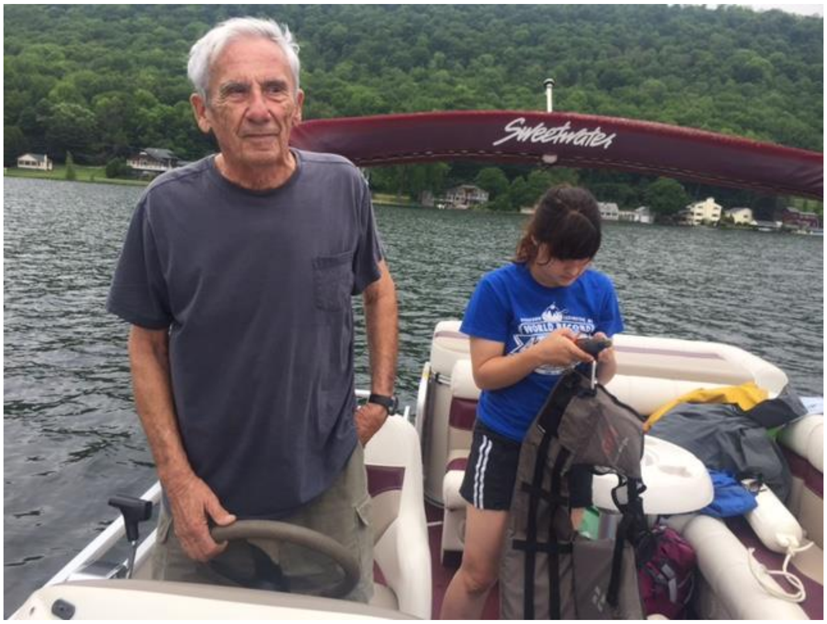
2018 Project Report:
Update on the Impact of Walleye Stocking and Milfoil Herbivorous Insects on the Growth of Eurasian watermilfoil in DeRuyter Reservoir



31 March 2019

Milfoil monitoring team, John Kennedy provided transportation for 2017 sampling. Photo by A. Barber

Paul H. Lord
paul.lord@oneonta.edu

SUNY Oneonta Biological Field Station
5838 State Highway 80
Cooperstown, New York 13326

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DeRuyter Milfoil tips: minimally damaged (left) and extensively damaged (right). Photos by L. Adams

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(Attn: Carol Jeschke)

Authors

Paul H. Lord ` Sarah Coney
Paul.lord@oneonta.edu sarah.coney@oneonta.edu
lordp@usa.net

SUNY Oneonta Biological Field Station
5838 State Highway 80
Cooperstown, New York 13326

Summary

The Tioughnioga Lake Association, the Tioughnioga Lake Preservation Foundation, Inc., and Madison County have long sought to manage Eurasian watermilfoil (*Myriophyllum spicatum*) hereinafter referred to as “milfoil,” because we found no native milfoils persisting in DeRuyter Reservoir. Milfoil management facilitates lake recreational uses that are obstructed by milfoil’s aggressive, invasive growth. This report summarizes the work of the 2018 cooperative effort of the Tioughnioga Lake Preservation Foundation, Inc., the Tioughnioga Lake Association and the Biological Field Station, SUNY Oneonta (BFS, Cooperstown). Prepared for Tioughnioga Lake Preservation Foundation and the Tioughnioga Lake Association, it presents the 2018 DeRuyter Reservoir data collected, its analysis, and offers recommendations on milfoil management and research.

<u>Key conclusions</u> include:

- Milfoil growth has not increased or decreased throughout the Reservoir
  - with the most recreational impeding milfoil at the south end
- DeRuyter Reservoir holds a modestly diverse submersed plant community
  - 20 aquatic plant and macroalgae species as of 2018
- Biocontrol of milfoil was no better in 2018 than in 2017
- Silt deposition from tributaries may be facilitating milfoil growth
  - particularly in southern end of Reservoir.

<u>Recommendations</u> include:

- Stocking with 45,000 walleye (*Sander vitreus*) fingerlings
  - to meet our recommended stocking rates in addition to adjusting for small sizes stocked in 2015 and low numbers in 2017 and 2018
  - to ensure consistent suppression of insect eating fish
  - to facilitate diversity of milfoil herbivores
- Continued monitoring of DeRuyter Reservoir milfoil, herbivores, algal blooms, and fish including June and autumn electrofishing
  - to evaluate stocking impacts
  - to adjust stocking in subsequent years.

<u>Content Organization</u>

The following three contents pages describe the organization of this report. Tables and figures herein are referred to as “Table 1, Table 2...” and “Figure 1, Figure 2…” respectively if they are contained in the main report while tables and figures contained in the appendices are referred to with the letter assigned to that appendix (*e.g*., “Figure A1,” “Table B1,” *etc*.). The main report is numbered 1 through 42 with appendices numbered with a letter, a dash, and the appendix page number (*e.g*., “B-3”).

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Background

Biological Field Station, SUNY Oneonta and DeRuyter Reservoir

The Tiougnioga Lake Association, the Tioughnioga Lake Preservation Foundation, the Onondaga County Health Department, and the Madison County Planning Department face a continuing challenge in their goal of suppressing Eurasian watermilfoil (*Myriophyllum spicatum*) to facilitate recreation on DeRuyter Reservoir (aka DeRuyter Lake) while minimizing unintended ecological consequences to DeRuyter Reservoir. Our 2018 research supported achievement towards that goal.

Our Biological Field Station, SUNY Oneonta (BFS, Cooperstown) research focuses on developing protocols for the biological control (biocontrol) of this noxious exotic aquatic plant. (Appendix B [p. B-1] provides definitions for technical terminology.) Previous BFS, Cooperstown work in Madison County demonstrated that augmented populations of milfoil herbivores (*e.g.*, Madison County's Lake Moraine 1998; 2000 and Lebanon Reservoirs 2001) do not necessarily have lake-wide milfoil-reducing impacts while work in Chautauqua Lake (2004 – 2012) demonstrated that augmented populations in some lakes can have an impact that spreads from the point of augmentation (unpublished data). Additional research in 2003 involving eight Madison County lakes, established a connection between sunfish (*Lepomis* sp.) numbers and the numbers of an important milfoil insect herbivore, the aquatic macrophyte moth (*Acentria ephemerella;* Figure 1) which has been confirmed by work in Minnesota (Ward & Newman, 2006). Moreover, aquatic macrophyte moth populations were associated with reduced Eurasian watermilfoil density (Lord, 2004).

Figure 1. Aquatic macrophyte moth larvae found in 2016 DeRuyter Reservoir sampling. Photo by A. Barber

DeRuyter Reservoir is a moderately productive (mesotrophic) lake of approximately 557 acres (CSLAP DeRuyter Reservoir, 2016) located in Madison County (Towns of DeRuyter and Cazenovia) and in Onondaga County (Town of Fabius) (Delorme, 1998). DeRuyter Reservoir is an artificially deepened lake with a maximum depth of approximately 53 feet (16m) and good water clarity (CSLAP DeRuyter Reservoir, 2016) facilitating rooted plant growth to depths of 18 feet which encompasses approximately 55% of the Reservoir (our estimate). Recreational uses of DeRuyter Reservoir are the focus of concern in regard to algae and plant growth with the result that the Reservoir is "listed" in the NYSDEC Priority Waterbody Listing for the Susquehanna River (NYSDEC PWL, 2015; CSLAP DeRuyter Reservoir, 2016).

The purpose of our 2018 effort in DeRuyter Reservoir was to monitor milfoil presence and density, milfoil herbivores, and fish community changes in DeRuyter Reservoir in the aftermath of walleye fingerling stocking. We made collections of milfoil plant stems to ascertain the presence or absence of milfoil herbivores and the impact, if any, that they were having on the Eurasian watermilfoil. Concurrent with our mapping of Eurasian watermilfoil, we mapped native aquatic plants found in the DeRuyter Reservoir. Herein, we report DeRuyter Reservoir physical and chemical data collected by SUNY Cobleskill. Additionally, SUNY Cobleskill electrofished the Reservoir in June and October to determine numbers of sunfish so that we could consider what impacts sunfish are having on milfoil herbivores. Figure 2 (modified from Lord *et al*, 2004) provides a summary of supporting research and our current working hypothesis in regard to fish community impacts on milfoil herbivores and Eurasian watermilfoil (Lord, 2004; Lord *et al*., 2004).

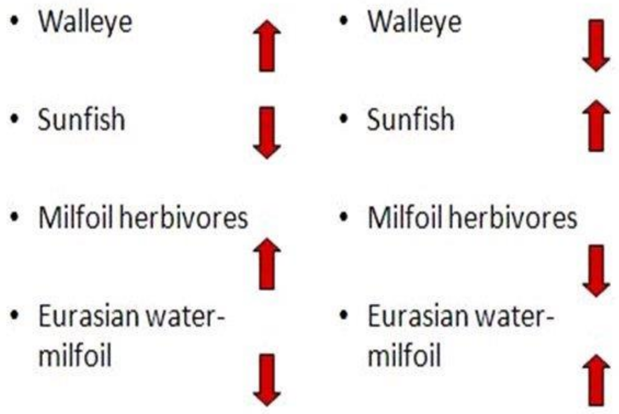


Figure 2. Hypothetical model illustrating understood trophic relationships between walleye (*Sander vitreus*), bluegill and pumpkinseed sunfish (*Lepomis* spp.), milfoil insect herbivores, and Eurasian watermilfoil (*Myriophyllum spicatum*) as perceived from NY State lakes data. Columns represent contrasting walleye and Eurasian watermilfoil population sizes. Up arrows represent larger populations whereas down arrows represent smaller populations.

Eurasian watermilfoil and Control Methods

Eurasian watermilfoil (*Myriophyllum spicatum*) is an exotic species believed to have been introduced into North American waters in the early 1880s near the Chesapeake Bay (Reed, 1977), although Couch and Nelson (1985) make a good case that it may not have been introduced until the early 1940s. Aiken *et al*. (1979) authoritatively describes Eurasian watermilfoil. Eurasian watermilfoil is difficult to distinguish from other members of its genus (close relatives) native to North American (Aiken *et al*., 1979; Aiken, 1981; Center, 1981; Gerber & Les, 1994) as noted by a review of Crow and Hellquist (2000), which differentiates members of the genus by flower parts. This is unfortunate because, in many Northeastern U.S. lakes, watermilfoil species flowers are evident for short periods, if at all (personal observation). Compounding the identification challenge, Eurasian watermilfoil can hybridize with native milfoils (Moody & Les, 2003; Thum, 2003; Pullman, 2006, Zuellig *et al*., 2012; Marko & Newman, 2018). Species identification of Eurasian watermilfoil, when not in flower, is based on the relative flaccidity and number of the dissected veins that form the leaves, the acuteness of the leaf, and the depth at which the plant flourishes. Eurasian watermilfoil typically has more than eleven dissected veins on each side of the leaf (Crow & Hellquist, 2000); however, care needs to be exercised in using this guideline since Eurasian watermilfoil can have between five and 25 vein pairs (Aiken, *et al*. 1979). Unlike local native watermilfoils, Eurasian watermilfoil's dissected veins appear flaccid and lay against the stem when the plant is removed from the water and held upside down. Additionally, Eurasian watermilfoil branches copiously as its stem approaches the water's surface, while natives rarely branch in water greater than 3 feet deep. Finally, several of our native watermilfoils produce turions for overwintering, which are evident in late autumn, winter, and early spring, while Eurasian watermilfoil and its hybrids often persist as a perennial evergreen herb (Aiken *et al*., 1979; Grace & Wetzel, 1978, personal observation).

Eurasian watermilfoil lives under the ice, growing when light is available (under clear ice), and dying back slowly toward its root crown (under snow covered ice) when light is not available (personal observation; Aiken *et al*., 1979). When the ice melts, root crowns (or stolons: horizontal stems from the plant base that produce new vertical stems) sprout new stems and persisting stems grow quickly, buoyed by stored carbon dioxide and dominating native plants that need warmer water for early season growth (Titus & Adams, 1979a; Creed & Sheldon, 1994; personal observation). If Eurasian watermilfoil reaches the water's surface without interference, it quickly forms dense canopies shading out most other aquatic plants beneath it and the lower leaves on its own stem, which slough off (Grace & Wetzel, 1978; Titus & Adams, 1979a; Aiken *et al*., 1979; personal observation). Flowers are sometimes formed, typically in July or August in the Northeastern U.S. Seed production may follow. Reproduction, however, is usually asexual, by expansion of root crowns and by fragmentation (Aiken *et al*., 1979; Madsen, 1993; Madsen 1998).

A variety of authors (Grace & Wetzel, 1979; Aiken *et al*., 1979; Titus & Adams, 1979b; Smith & Barko, 1990; Madsen, 1993) describe a form of asexual reproduction in Eurasian watermilfoil: abscission or autofragmentation. Healthy Eurasian watermilfoil may not fragment itself; rather, long stems, particularly ones that form canopies, are subject to twisting, weakening or breakage. Weakened stems interrupt the flow of nutrients from roots to plant tips (meristems). This can occur by wind driven wave energy or by animal or human entanglement or by the actions of herbivores feeding on Eurasian watermilfoil. Regardless of how Eurasian watermilfoil is fragmented, roots quickly develop on viable pieces not well connected to stem and roots. A change in buoyancy accompanies this development. Stem fragments without roots are positively buoyant (Grace & Wetzel, 1978), and, as a consequence, gain better access light, while rooted fragments become negatively buoyant (Grace & Wetzel, 1978), affording better access to substrates. Eurasian watermilfoil obtains two frequently limiting nutrients, phosphorus (P) and nitrogen (N), from the substrate (Best & Mantai, 1978; Painter & McCabe, 1988; Lillie & Barko, 1990; Brade & Mantai, 1991) although it obtains ammonium (also N) from the water (Wetzel, 1983; Walstad, 1999).

Eurasian watermilfoil, now found throughout North America, inhabits a wide variety of lakes, but is most dense in moderately productive (mesotrophic) and excessively productive (eutrophic) lakes (Smith & Barko, 1990; Madsen, 1998). When abundant, Eurasian watermilfoil impedes boating, water-skiing, fishing, and swimming, by forming a thick surface canopy (Smith & Barko, 1990). Development of protocols using insect herbivores to control this noxious, exotic aquatic macrophyte has been noted to be a worthwhile objective by various researchers (Batra, 1977; Buckingham & Ross, 1981; Kangasniemi, 1983; Oliver, 1984; Painter & McCabe, 1988; MacRae *et al*., 1990; Creed *et al*., 1992; Sheldon & Creed, 1994; Sheldon & Creed, 1995; Perry & Penner, 1996; Newman *et al*., 1997; Johnson *et al*., 1998; Cofrancesco & Crosson, 1999; Newman *et al*., 2002; Johnson & Blossey, 2002; Tamayo, 2003). However, other methods of control have also been used.

Physical methods of elimination were first used to control Eurasian watermilfoil (Cofrancesco, 1998) and they remain in use today. Mechanical harvesting, which removes nutrient laden plant material, has been used widely with some unanticipated results. If not cut repeatedly over the course of a season, Eurasian watermilfoil often re-grows denser after cutting. Insects inhabiting the upper portions of the plants (including milfoil herbivores) are killed. Four percent or more of the fish, largely young-of-the-year bluegill, are killed in harvested areas and game fish habits change (Nichols & Cottam, 1972; Mikol, 1985; Engle, 1990; Valley & Bremigan, 2002a,b) unless extraordinary attention is focused on saving fish. Harvesting does remove recreationally impeding macrophyte stems while leaving other macrophyte plant parts rooted in the substrates stabilizing the lake bottom and providing refuge areas for invertebrates and small fish. Nutrients appear to be pumped from the sediments via these cut stems (Bossong *et al*., 2019) potentially facilitating algae blooms.)

Hand pulling and substrate barriers have been successfully employed in some North American lakes to keep nascent Eurasian watermilfoil infestations from developing further. The key to satisfactory employment of these techniques appears to be early detection and action (LaMere, 1999; Smagula, 2002). Winter drawdown of water levels in lakes with controllable water levels has been widely used to freeze and kill Eurasian watermilfoil in shallow areas. A properly timed drawdown and winter reflooding can remove organic sediment from the littoral zone increasing the average size of particles in the substrate and reducing Eurasian watermilfoil densities for subsequent growing seasons (Lyman, 2001). However, such drawdowns may also make significantly more phosphorus available for algae and surviving macrophytes (Klotz & Linn, 2001), while killing nontarget organisms (*e.g.*, invertebrate animals).

Inorganic chemical control approaches to aquatic macrophyte control were attempted shortly after physical methods were explored. In the 1940s, complex synthetic carbon compounds were first used (Cofrancesco, 1998). Six chemicals (available in a wide variety of formulations not all of which are appropriate for aquatic use) used in the U.S. to target Eurasian watermilfoil can permit "selectivity to target milfoil" allowing some portion of the native plant community to persist. (Other chemicals with apparent selective effects are undergoing testing.) Results, in practice, with all of the approved "selective" chemicals have been varied with regard to their efficacy in reducing Eurasian watermilfoil and their impacts on desired native species.

Flumioxazin (2-[7-fluoro-3,4-dihydro-3-oxo-4-(2-propynyl)-2H-1,4-benzoxazin-6-yl]-4,5,6,7-tetrahydro-1H-isoindole-1,3(2H)-dione) is the newest herbicide approved for lakes in New York State. It is sold as Clipper® by Valent U.S.A. Corporation. It is PPO inhibitor which initiates cell membrane disruption (Britton, 2011; Valent U.S.A. Corp.).

Imazamox (2-[4,5-dihydro-4-methyl-4-(1-methylethyl)-5-oxo-1H-imidazol-2-yl]-5-(methoxymethyl)-3-pyridinecarboxylic acid) is a selective herbicide approved for lakes in New York State. It is sold as Clearcast® by Sepro Corporation, and it is an imidazolinone pesticide. It interferes with a plant biosynthesis pathway for creating three amino acids (leucine, isoleucine, and valine). It is effective against Eurasian watermilfoil, but it impacts many native plants at dosages less concentrated than those needed to control milfoil (100 – 200 ppb). In water exposed to sunlight, Imazamox degrades quickly, but it can persist in aquatic sediments (PIMS, 2011; SePRO, 2010).

Triclopyr (3,5,6-trichloro-2-pyridinyloxyacetic acid combined with triethylamine salt [$C_7H_4Cl_3NO_3$]) was approved for use by EPA (Poovey *et al*., 2000; SePRO, 2003a; Mongin, 2003), and it is now authorized for use in New York. Triclopyr is sold as Renovate 3®, and Garlon 3A®. It is a systemic selective herbicide, which kills plants by mimicking a plant hormone (auxin) causing unsustainable growth (SePRO, 2003a). Considerable uncertainty is still associated with the selectivity of Triclopyr documented in Getsinger *et al*., 1997 for a riverine situation (not in a lake) with a wide variety of aquatic plants (Hyde, 2004). Some natives of concern; waterweed (*Elodea canadensis*), thin leaved pondweed (*Stuckenia pectinata=Potamogeton pectinatus*), and water celery (*Vallisneria americana*) are reportedly quite tolerant of this herbicide (Sprecher & Stewart, 1995).

2,4-D (2,4-diclorophenoxy acetic acid [$C_8H_6Cl_2O_3$]) use against and impact on Eurasian watermilfoil is described by Aiken *et al*. (1979) and Gangstad (1982). 2,4-D is the longest used "selective" aquatic herbicide. It is still widely used and marketed as Aqua-Kleen®, DMA 4, Navigate and Weedar® (Hoyer & Canfield, 1998; Vermont Department of Environmental Conservation, 1993; Gallager, 1992; Humberg *et al*., 1989). 2,4-D is a pseudo plant hormone (auxin) that resists counteraction by its controlling hormone (IAA oxidase) forming proteins that block plant stem flows and prevent energy storage (Levitt, 1969). It is somewhat selective for Eurasian watermilfoil (Vermont Department of Environmental Conservation, 1996).

Fluridone (1-methyl-3-phenyl-5(3-(trifluromethyl) phenyl)-4(1H)-pyridinone [$C_{19}H_{14}F_3NO$]) is marketed as Sonar® with several formulations available by SePRO ® and was previously marketed as Avast!^{TM} by Griffin L.L.C.$^{®}$. (SePRO Corporation$^{®}$, 2003b; Griffin L.L.C.$^{®}$, 2003; SePRO Corporation, 2004). It is currently the most aggressively marketed "selective" aquatic herbicide. It acts by interfering with carotenoid production causing plants to die from sunlight that breaks up chlorophyll molecules throughout the plant. Its impact is long-lasting and any product not absorbed by plants is broken down by light and microbial action (Hoyer & Canfield, 1998; Vermont Department of Environmental Conservation, 1996; SePRO Corporation$^{®}$, 2003b; Griffin L.L.C.$^{®}$, 2003). This product has been used, with selective dosages (~ 6 ppb) and selective results, in the south basin of Lake Moraine, Madison County since 1996 and in the north basin in 2004 (Harman & Albright, 1997; Harman, *et al*., 1998; 2000a; 2001a; 2002; 2004; Albright, 2004). It has also been used, with little apparent selectivity, in Waneta Lake (Madsen *et al*., 2001; Lord *et al*., 2005; Johnson, *et al*., 2006).

Endothall (3,6-endoxohexahydrophthalic acid [$C_8H_{10}O_5$]) is available as a dipotassium compound, a disodium compound and as an amine salt of endothall. It is sold as Aquathol$^{®}$ K, Aquathol$^{®}$ Super K and Hydrothol$^{®}$ 191 by Cerexagri, Inc. (Cerexagri, Inc., 2004, Hoyer & Canfield, 1998; Elf Atochem$^{®}$ North America, Inc, 1996). It works as a contact herbicide that is somewhat selective for Eurasian watermilfoil (Netherland, 1991; Oregon State University, 1998; Vermont Department of Environmental Conservation, 1996). The product is a pseudo-hormone that promotes ethylene production (Abeles, 1973), which stops fat and protein synthesis and inhibits respiration (MacDonald *et al*., 1992). Endothall not absorbed by plants is broken down by the metabolism of microbes (Elf Atochem$^{®}$ North America, Inc, 1996).

The current list pesticide chemicals approved for use in New York is found on the NYSDEC web site for "Bureau of Pest Management - Information Portal" which can be found at http://www.dec.ny.gov/nyspad/?0. To locate the sub list of chemicals approved for aquatic situations, click on "Advanced Search" and in the "Restriction" field select "Class H (Surface water application restricted)" and in the "Registration Status" field select "Registered" and click "Search."

Many lake property owners are intuitively reluctant to use chemicals in their lakes. Since all of the above described chemicals interfere with metabolic processes and stress lakes by stressing most, if not all, of the plants in them (while killing only some of them), some lake property owners are reluctant to use chemicals to control Eurasian watermilfoil (personal observation).

In the late 1950s, the U.S. government started a program evaluating biocontrol organisms for aquatic macrophytes (Cofrancesco, 1998), and in the 1960s two diseases (northeast disease and Lake Venice disease) were linked to Eurasian watermilfoil declines (Painter & McCabe, 1988). Bacterial and fungal pathogens of Eurasian watermilfoil have been tested and some have shown promise, but exotic pathogens have yet to be released, while native ones do not appear to be able to control Eurasian watermilfoil without some other Eurasian watermilfoil weakening agent (Shearer, 2000; Shearer, 2002, Hussner et al., 2018). The exotic, but nonreproductive, triploid hybrids of grass carp (*Ctenopharyngodon idella* Valenciennes [Cypriniformes: Cyprinidae]) have been used in many locations to control Eurasian watermilfoil, but this fish is not a Eurasian watermilfoil-specific herbivore. Moreover, it tends to eat other plants preferentially to Eurasian watermilfoil in the northeast (NYSFOLA, 2009; Lynch 2009). Its use in the northeastern U.S. is typically restricted to isolated impoundments (generally farm ponds) with little chance of carp movement downstream (Pine & Anderson, 1991; Kirk, 2000; Madsen, 2000; Pipalova, 2002; Lubnow *et al*., 2003). Not surprisingly, Eurasian watermilfoil biocontrol articles focus on milfoil insect herbivores.

The yearly cycle of Eurasian watermilfoil growth, reproduction, and persistence under the ice is facilitated by carbohydrate storage in the root crowns (AKA stolons) (Titus & Adams, 1979b). Madsen (1993) suggests targeting control attempts to those periods when stored carbohydrates are minimal. Unfortunately, as he, and Titus and Adams (1979b) note, carbohydrate storage and usage patterns vary widely from year to year and from site to site.

Eurasian watermilfoil is reportedly not a management problem in its native range (Center, 1981; Smith, 1982). Spencer and Lekic (1974) reported "…25 insect species feeding on [Eurasian watermilfoil]." Aiken *et al*. (1979) noted "…no insect parasites [herbivores of milfoil] have been reported in North America." That has changed (Newman, 2004). Although some authors discount their potential (Center, *et al*., 2002), three North American insects are now identified as having some potential to control Eurasian watermilfoil: *Acentria ephemerella*, an aquatic macrophyte moth; *Cricotopus myriophylli*, the milfoil midge; and *Euhrychiopsis lecontei*, the milfoil weevil (Kangasniemi *et al*., 1992; Johnson *et al*., 1998; Newman *et al*., 2002; Johnson & Blossey, 2002, Newman, 2004). Additionally, we have noted that other midges damage Eurasian watermilfoil by their overwintering activities and stem mining (as have others [Kangasniemi & Oliver,1983; MacRae & Ring, 1993]), but not to the extent that they normally have the potential for meaningful control, while other researchers have noted similar impacts from other aquatic macrophyte moths (Batra, 1977; Buckingham & Ross, 1981; Newman *et al*., 1999; Newman *et al*., 2002).

Aquatic Macrophyte Moth: *Acentria ephemerella* (Denis & Schiffermüller) (Lepidoptera:Pyralidae) (= *Acentria nivea* Olivier; Passoa, 1988).

Batra (1977) first noted an insect in the U.S. with Eurasian watermilfoil herbivory potential: the exotic aquatic macrophyte moth (*Acentria ephemerella*, hereinafter referred to as "the moth" or "moth larvae", as appropriate). She described the moth and its behavior in all life stages and its impact on Eurasian watermilfoil in the field and under laboratory conditions, whereas Berg (1942) provided the most extensive documentation of this moth in its native habitat. Batra concluded that it might have potential for control of Eurasian watermilfoil since it damaged stems and ate leaves, but that host preference testing was needed. Buckingham & Ross (1981) provided no-choice testing on a variety of aquatic plants and concluded that the moth was not specific to Eurasian watermilfoil and that its dislike of algae-covered Eurasian watermilfoil limited its potential. Painter & McCabe (1988) found that eight larvae per ten tips had a severe impact on Eurasian watermilfoil in a laboratory experiment and could control Eurasian watermilfoil. They also found in the Kawartha Lakes, where Eurasian watermilfoil declined by 95%, moth larvae in the same abundance (six larvae per ten apical stems 10 in; 25 cm), as reported in the Cayuga Lake Eurasian watermilfoil decline (Johnson *et al*., 1998). Cornell University work (Johnson *et al*., 1998; Gross *et al*., 2000; Johnson *et al*., 2000) shows that the moth will eat other aquatic macrophytes, but it prefers Eurasian watermilfoil and that its presence has facilitated the return to dominance of waterweed in southern Cayuga Lake and the return to dominance of water celery and water stargrass (*Heteranthia dubia*) in northern Cayuga, NY (Zhu & Georgian, 2014). The moth damaged apical meristems preventing Eurasian watermilfoil from reaching the water's surface where it could shade out competing native aquatic macrophytes. Further, Gross *et al*. (2001) contends that the moth along with the milfoil weevil was responsible for Eurasian watermilfoil control attributed to the milfoil weevil in a variety of other studies.

The moth overwinters as a larva in Eurasian watermilfoil and other plant stems and leaves, or on the growing tips of coontail (*Ceratophyllum demersum*). Larvae may have as many as five instars with the first instar burrowing into a single dissected vein of Eurasian watermilfoil and a second instar forming a shelter, known as a "refuge", out of two adjoining dissected veins. Later instars form even larger refuges out of more dissected veins or entire leaves. In the spring, many larvae end up in the organic debris comprised of decaying aquatic macrophytes from the previous season. They become active at about 10^o C (although some are active in cooler water in certain situations). They eat growing meristems and leaves of Eurasian watermilfoil and later instars build refuges out of Eurasian watermilfoil leaves. Larvae are almost always found within 25 cm of the apical meristem. Final instar larvae pupate in shelters comprised of leaf and stem material adhered to the stem. After pupation, the adults swim to the surface. The males have wings capable of flight while the females normally possess flightless, vestigial wings and float on the surface. Females emerge at night and swim about on the water's surface whereas males, resting on surfaces near the water during the day, fly just above the surface at dusk and at night searching for females (Berg, 1942, Batra, 1977; Painter & McCabe, 1988; Johnson & Blossey, 2002). Mating takes place in seconds when a male finds the female swimming with posterior poised above the water's surface (Buckingham & Ross, 1981; Johnson, 2000). After mating, the female swims down one meter or more and lays eggs along Eurasian watermilfoil leaves. Adults rarely live more than 48 hours while the larvae take from two to eleven months to develop when eating Eurasian watermilfoil. Although there appears to be no more than two generations of moths per season in the North America, some adult moths do emerge every month of the summer, apparently synchronized by moon phases with peak emergences at the end of June and in mid-August (Berg, 1938; Berg, 1942; Lange, 1956; Buckingham & Ross, 1981; Palm, 1986; Painter & McCabe, 1988; Johnson, 2000; Johnson & Blossey, 2002; personal observation). Figure 2 is a photo of an Aquatic macrophyte moth larvae found in DeRuyter Reservoir in 2016.

European populations of Eurasian watermilfoil have been investigated for their direct and indirect chemical defenses against moth herbivory (Gross & Bakker, 2012; Gross *et al*., 2002; Choi, *et al*., 2002; Leu, *et al*., 2002; and Walenciak, *et al*., 2002). Eurasian watermilfoil does produce chemicals when grazed upon by the moth and the moth avoids eating Eurasian watermilfoil with those chemicals (Fornoff & Gloss, 2014).

<u>Milfoil Weevil</u>: *Euhrychiopsis lecontei* (Dietz) (Coleoptera: Curculionidae)

Following the initial research of Sheldon and Creed (Creed *et al*.,1992; Creed & Sheldon, 1994; Creed & Sheldon, 1995; Sheldon & Creed, 1995), most Eurasian watermilfoil herbivory research has focused on the watermilfoil weevil (*Euhrychiopsis lecontei*) (*e.g*., Newman *et al*., 1996; Sheldon & O'Bryan, 1996a,b; Solarz & Newman,1996; Hutchinson, 1997; Newman *et al*., 1997; Sheldon, 1997a,b; Sutter & Newman,1997; Jester, 1998; Tamayo, 1998; Cofrancesco & Crosson, 1999; Jester & Bozek, 1999; Mazzei *et al*., 1999; Tamayo, *et al*., 1999; Creed, 2000a,b; Jester *et al*., 2000, Lillie, 2000; Newman & Biesboer, 2000; Tamayo, *et al*., 2000; Newman *et al*., 2002; Johnson & Blossey, 2002; Tamayo, 2003, Parsons, *et al*., 2011; Newman, 2012, Borrowman *et al.*, 2014, Hovel *et al*., 2017, Roketenetz *et al.*, 2017). The watermilfoil weevil not only eats Eurasian watermilfoil, but it destroys the buoyancy of Eurasian watermilfoil causing stems to drop to the bottom. This interferes with Eurasian watermilfoil's competitively advantageous ability to form a canopy, which shades out other plant species (Creed *et al*., 1992; Creed & Sheldon, 1995; Sheldon & Creed, 1995). Creed and Sheldon (1995) note that the milfoil weevil evolved on a diet of native watermilfoils and suggest that its annual life cycle might be out of synchrony with the exotic Eurasian watermilfoil. EnviroScience® Incorporated's MiddFoil™ process augmented local populations of the milfoil weevil and was based on the research of Sheldon & Creed (Hilovsky, 1998; Hilovsky, 2000, Hartzel, 2003, Envisoscience, 2014). The milfoil weevil prefers Eurasian watermilfoil to native milfoils (Sheldon & Creed, 1995) and produces more eggs and develops faster on a diet of Eurasian watermilfoil (Newman *et al*., 1997; Marko & Newman, 2017).

The milfoil weevil overwinters as an adult in soils and in organic litter (Creed & Sheldon, 1994; Cofrancesco & Crosson, 1999; Johnson & Blossey, 2002; Thorstenson *et al.,* 2013) adjacent to lakes and ponds with Eurasian watermilfoil and/or native milfoil species. A lakeshore buffer of 15'-40' with fallen leaves and uncut grass facilitates overwintering (et al., Thorstenson *et al.,* 2013). When rising ice-free waters flood the winter habitat or when rising temperatures prompt their return, the adults migrate back to the water and feed on recently formed leaves (personal observation). After a week or so, the adults mate and, shortly thereafter, the female starts laying an egg or two a day, on meristem material (Creed & Sheldon, 1995; Cofrancesco & Crosson, 1999). After three to five days, newly hatched larvae burrow into the stem immediately below the meristem releasing stem tissue (aerenchyma) gas that slowly changes Eurasian watermilfoil's buoyancy from positive to negative (Creed *et al*., 1992; Newman *et al*., 1996). As the larvae grow, they eat increasingly larger diameter tunnels through the stems. Larvae pupate in the stems and emerge after nine to twelve days to begin a new generation after a period of eating. The weevil's generation time, under ideal conditions of food and temperature, is 28 days (Newman *et al*., 1996; Cofrancesco & Crosson, 1999; Mazzei *et al*., 1999; Newman, 2000).

Adult milfoil weevils appear to have difficulty swimming unassisted to depths greater than 10 inches (personal observation). They also have no alternative food source when watermilfoils are not available, establishing a classic predator-prey population cycle (Smith, 1996; Jester, *et al*., 2000; Johnson, in prep). Additionally, adult milfoil weevils are at risk to fish predation when swimming from plant to plant, as they need to, for mating and egg laying (Sutter & Newman, 1997; Hairston *et al*., 2001; Cornwell, 2001, Maxson 2016; Newman, 2017). Additionally, predation by bluegill (*Lepomis macrochirus*) on Milfoil weevils is documented by Maxson (2016).

Milfoil Midge: *Cricotopus myriophylli* (Oliver) (Diptera: Chironomidae)

Least studied of the three acknowledged herbivores with Eurasian watermilfoil control potential is the milfoil midge (*Cricotopus myriophylli*). Kangasniemi and Oliver (1983) noted that the milfoil midge caused significant damage to Eurasian watermilfoil by using apical meristems for food, refuge construction, and pupating sites. Few U.S. researchers have reported finding the milfoil midge (Newman & Maher, 1995; Johnson *et al*., 2000; Johnson & Blossey, 2002; Lord, 2004), possibly reflecting the difficulty of observing and identifying these small insects. Kangasniemi (1983) noted the milfoil midge had "potential for use as a biocontrol agent." Further research (MacRae *et al*., 1990; Kangasniemi, *et al*., 1992) led to the conclusion that with sufficient numbers of milfoil midges (approximately one per apical meristem), milfoil overall height could be reduced and the plants prevented from surfacing and flowering even while little plant biomass was consumed.

Milfoil midges are small and their larvae are easily overlooked even when using a stereoscopic dissecting microscope to examine milfoil (personal observation). This is particularly true with early instars. The milfoil midge is not listed in the definitive key of the *Cricotopus* genus in the region (Simpson, *et al*., 1983). Definitive identification involves clearing or crushing head capsules and looking at mouth parts (Oliver, 1984). Other diagnostic methods may suffice seasonally in local situations because of limited local midge diversity (Berg, 2002). Milfoil midge eggs have not been located in the field, although they have been collected in laboratory cultures. Milfoil midges overwinter in Eurasian watermilfoil meristems in dormancy as 2nd, 3rd, and 4th instars, and become active with water temperatures of 10 - 15°C. Four larval instars and a pupae stage precede adulthood. Swarms of adults have been noted flying approximately 10 feet above the water's surface. Milfoil midges complete a life cycle a year, but their emergence is not well synchronized. They can live on a diet of native milfoils leading to a belief that it is a native insect although a seemingly identical insect has been found in Europe (MacRae, 1999) and in Serbia (Kroutsova & Voilo, 2015). Milfoil midges may eat other plants when milfoils are not available, but show a preference for milfoils (MacRae *et al*., 1990; Kangasniemi, *et al*.; 1992; MacRae & Ring, 1993).

Other Herbivores

In Lebanon Reservoir, Madison County, NY, we have seen significant mining of milfoil basal (bottom) stems by a nonbiting midge of the genus *Glyptotendipes* sp. previously associated with Water stargrass (*Heteranthera dubia* [Harms, 2010]. In Chautauuqua Lake and in DeRuyter Reservoir we have two separate caddis fly larvae (*Nectopsyche albida* [Johnson *et al*., 2012]; *Leptocerus* sp. [Lord, per obs.]) that eat milfoil.

Herbivory Potential

Insect herbivores are unlikely to eradicate Eurasian watermilfoil in any waterbody (Hussner, 2017). However, insect milfoil herbivores do have the potential to keep Eurasian watermilfoil from impeding recreation by keeping it from reaching the water's surface, and, in some cases, herbivores can reduce its percentage of biomass in the plant community to single digits (personal observation).

Fish Predation

Numerous authors have speculated on the impact of fish predation on milfoil herbivores (Buckingham & Ross, 1981; Menzie, 1981; Jester, 1998; Creed, 2000; Lillie, 2000; Newman & Biesboer, 2000; Tamayo *et al*., 2000; Cofrancesco, 2000, Newman, 2012; Hussner, 2017). Some have even looked at fish behavior and/or population dynamics in the presence of herbivores in controlled situations (Newrough, 1993; Sutter & Newman, 1997; Cornwell, 2001; Hairston *et al*., 2001; Newman *et al*., 2002). Cornwell (2001) documented pumpkinseed sunfish (*Lepomis gibbosus*) predation on the milfoil weevil. Figure 3, modified from Hairston *et al*. (2001), provides insight. Lord (2004) and Lord *et al*. (2004) document an association between fish species population numbers and the numbers of the aquatic macrophyte moth (Figure 2). A Minnesota study concluded that “fish suppression of watermilfoil herbivores can have a positive, indirect effect on plant growth” (Ward & Newman, 2006) although recent studies have failed to produce a clear result (Newman, 2017) . More studies correlating the numbers of milfoil herbivores in Eurasian watermilfoil infested lakes with the numbers of fish of differing species, and within year-classes, need to be undertaken. Such associations appear to be important biological considerations affecting the ability of some herbivores to control Eurasian watermilfoil.

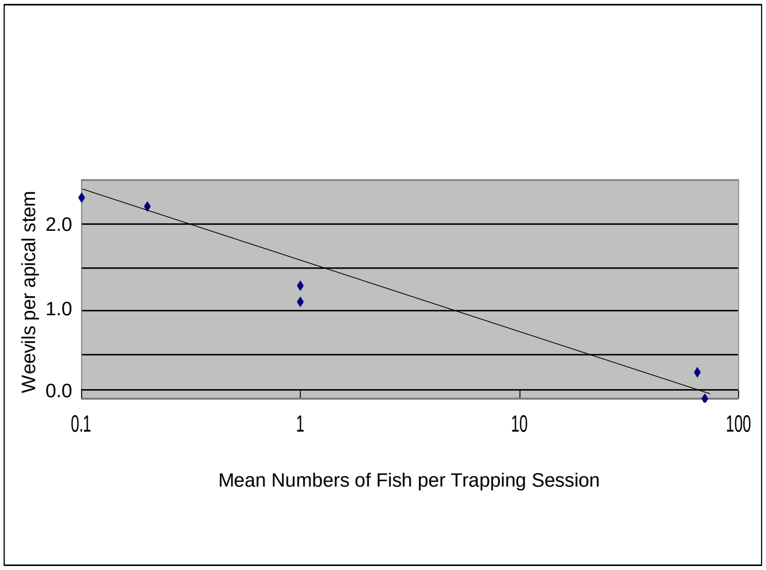


Figure 3. Graph modified from Hairston *et al*. (2001) illustrating inverse relationship found between fish numbers and milfoil herbivore numbers in Cornell University Research Ponds. Note the logarithmic scale of x-axis.

Other forms of herbivore predation must also be considered. Bats and even predacious invertebrates (*e.g*., dragonflies, damselflies, hydras, scuds, water mites, and flatworms) are possible limiting factors on herbivore numbers (Batra, 1977; Buckingham & Ross, 1981; Menzie, 1981). Additionally, the use of herbicides targeting Eurasian watermilfoil reduces the numbers of the Eurasian watermilfoil herbivores (Havel, *et al*., 2017).

Walleye are typically stocked as fry, June fingerlings or autumn fingerlings. Costs, size and survivability increase the longer the Walleye are held in culture prior to stocking in lakes.

Fry are normally provided free of charge in New York State. Fry are available in May and are approximately ¼" long. They are eaten by a wide variety of fish.

June fingerlings are grown in ponds rich with zooplankton which the fingerlings eat. Once the zooplankton are eaten, the June fingerlings must be stocked before they cannibalize each other. June fingerlings are normally 1⅓" to 1¾" long. They are eaten by Perch (*Perca flavens*), Rock bass (*Ambloplites rupestris*), Crappie (*Pomoxis* spp.) and game fish. In New York State, fingerlings must be tested for viral hemorrhagic septicemia and other diseases prior to stocking. Delays associated with disease testing frequently lead to higher water temperatures which stress the fingerlings and can lead to death of the fingerlings if moved.

Walleye kept in ponds after the zooplankton are eaten must be fed forage fish or they will cannibalize each other. In New York State, we typically feed pond raised walleye Fat-head minnows (*Pimephales promelas*) which, themselves must be raised in ponds. If fed plentiful Fat-head minnows, Walleye fingerlings will grow longer than 5½" by late September or early October, at which time pond water temperatures are cool enough to permit safe fingerling movement from ponds to the waterbody to be stocked. Autumn fingerlings can be eaten by longer game fish.

Land Use Impacts

The number of lawns, unpaved drives, wintertime salted paved roadways, and bare earth ditches surrounding DeRuyter Reservoir raises concerns regarding salt, fertilizer use and ditching. Fertilizers are effective in promoting growth in aquatic plants as well as terrestrial plants. Most of our North American lakes are limited in their algae and rooted plant productivity by phosphorous (P). Lawn fertilizers typically contain compounds comprised of three elements facilitating lawn growth: nitrogen (N), phosphorous (P), and potassium (K) which are listed on fertilizer labels in that sequence. These same elements are found in sewage and manure. P remains even after sewage has been processed in a standard septic system. Cornell University scientists have completed regular experiments in their Cornell University Research Ponds confirming the positive relationship between aquatic plant and algae growth and fertilizer use (Johnson, pers comm). Additionally, a number of other researchers have documented reduced productivity in lakes and ponds treated with chemicals that bind to the phosphorous in the water and sediments (Welch & Cooke, 1999).

Salt use facilitates Eurasian watermilfoil dominance. A review of the peer-reviewed scientific literature reveals that Eurasian watermilfoil grows vigorously at salinities up to 10% of sea salinity (~3.5 parts per thousand [ppt]) and can remain alive at 20% sea salinity (7 ppt) (Nichols & Shaw, 1986). Others maintain that Eurasian watermilfoil is even more tolerant of salt, surviving in waters with a salinity of 16 ppt (46% of sea salinity) and growing 15 ppt (~ 43% of sea salinity) (Reed, 1977). Some Eurasian watermilfoil can tolerate waters and still retain growing tips at 93% sea salinity, or about 32 ppt (Reed, 1977). Conversely, little is known about the salt tolerance of waterweed (*Elodea* spp.) or coontail. Other research describes many natives (*e.g*., water celery, water naiads [*Najas* spp.], and pondweeds [*Potamogeton* spp.]) that are less tolerant of salinity than Eurasian watermilfoil.

Little was known about macrophyte competitiveness under saline stressed conditions, but it is known that Eurasian watermilfoil, coontail, and waterweed typically flourish under similar conditions: at depth ranges of 3 feet to 13 feet (1 m to 4 m), temperatures ~86F (~ 30°C), and similar pH levels (Nichols & Shaw 1986; Reed 1977). In an experiment established in the Cornell University Research Ponds Muenscher Greenhouse (Lord *et al*., 2004b), Eurasian watermilfoil tolerated salinity causing adverse growth in native plants.

Methods & Rationale

Water Quality Sampling

SUNY Cobleskill sampled DeRuyter Reservoir water quality parameters on 26 June, 2018 at a deep location in the southern part of the Reservoir (Figure 4). This is not the deepest location, but it is the area consistently sampled at the time of electrofishing. The specific sampling site (18T 0426692 4741877 [UTM coordinates; NAD83 datum]) is depicted in Figure 4 as WQ2. Water quality was assessed using a multi-probe device calibrated per manufacturer's instructions, to measure water oxygen levels, pH, conductivity, and temperature.

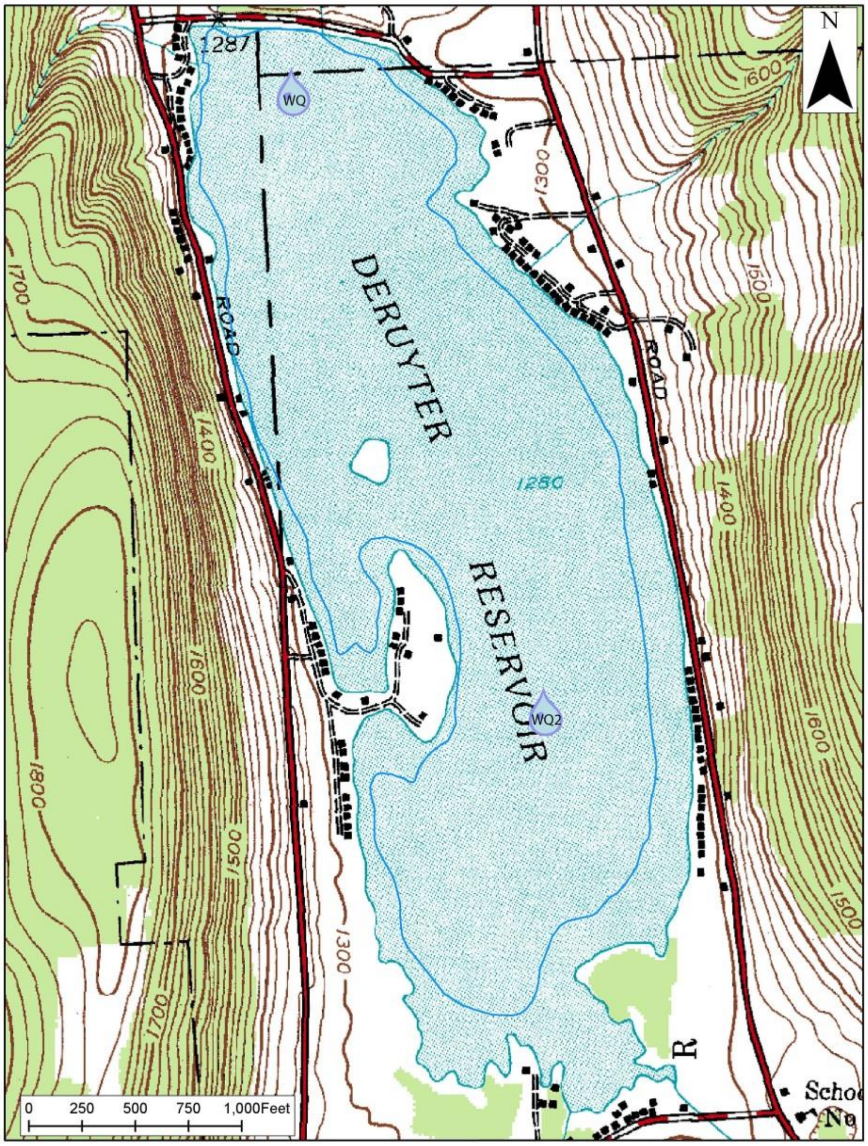
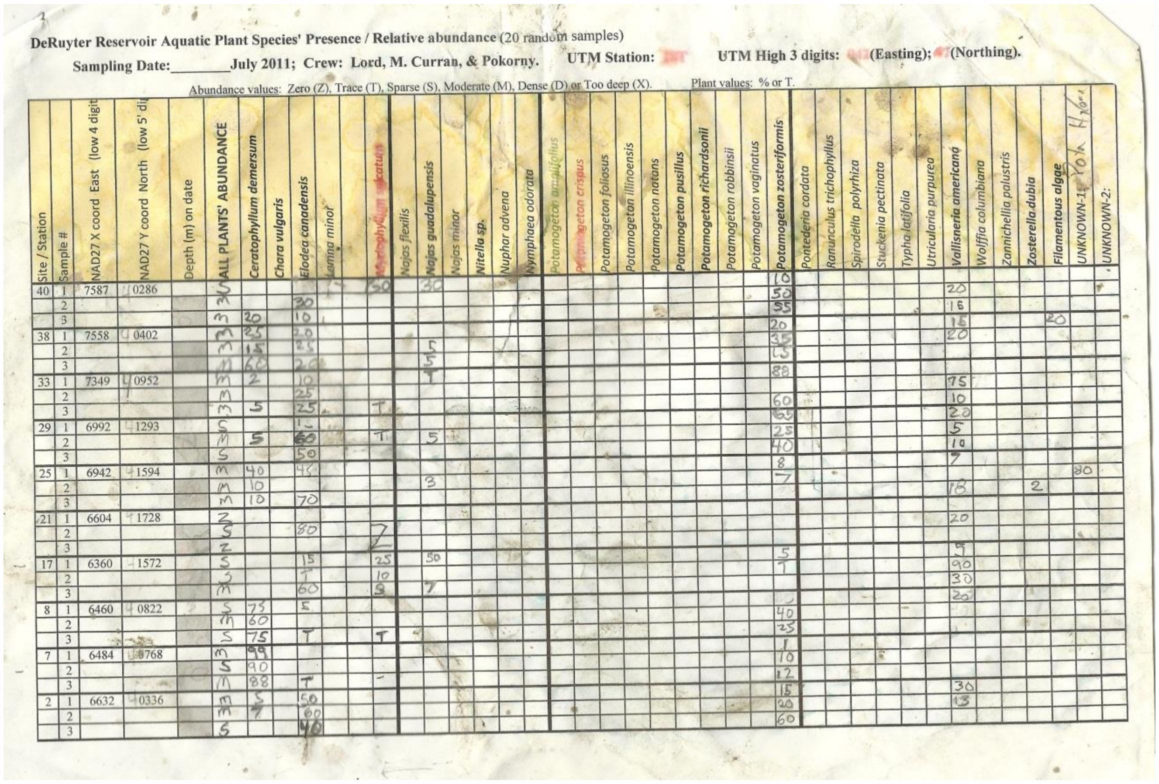


Figure 4. Location of DeRuyter Reservoir water quality sample (WQ2) obtained in 2018.

Macrophyte Sampling by the Point intercept rake toss relative abundance method (PIRTRAM)

Using a Garmin® mapping system (*e.g.*, Garmin® Inland Lakes Continental United States® [2008] and MapSource ® Version 6.16.3 [2010]), Universal Transverse Mercator (UTM) coordinates were plotted along the 10 foot depth interval in DeRuyter Reservoir to identify sampling sites (Figure 4). UTM coordinates were used because they facilitate sampling at precise distances of separation (Carnes, 2002). UTM coordinate information was entered in a sampling form (Figure 5). A handheld global positioning system (GPS) unit (Garmin GPSMAP 60CSx™) was loaded with sampling waypoints corresponding to the sites identified on the sampling form.



DeRuyter Reservoir Aquatic Plant Species' Presence / Relative abundance (20 random samples)

Sampling Date:________July 2011; Crew: Lord, M. Curran, & Pokorny. UTM Station: [illegible] **UTM High 3 digits:** [illegible] **(Easting);** [illegible] **(Northing).**

Abundance values: Zero (Z), Trace (T), Sparse (S), Moderate (M), Dense (D) or Too deep (X). Plant values: % or T.

Site / Station	Sample #	NAD27 X coord East (low 4 digit	NAD27 Y coord North (low 5' di	Depth (m) on date	**ALL PLANTS' ABUNDANCE**	***Ceratophyllum demersum***	***Chara vulgaris***	***Elodea canadensis***	*Lemna minor*		***Myriophyllum spicatum***	*Najas flexilis*	***Najas guadalupensis***	*Najas minor*	***Nitella sp.***	*Nuphar advena*	*Nymphaea odorata*	*Potamogeton amplifolius*	***Potamogeton crispus***	*Potamogeton foliosus*	*Potamogeton illinoensis*	*Potamogeton natans*	***Potamogeton pusillus***	***Potamogeton richardsonii***	*Potamogeton robbinsii*	*Potamogeton vaginatus*	***Potamogeton zosteriformis***	*Pontederia cordata*	*Ranunculus trichophyllus*	*Spirodella polyrhiza*	*Stuckenia pectinata*	*Typha latifolia*	*Utricularia purpurea*	***Vallisneria americana***	*Wolffia columbiana*	*Zannichellia palustris*	***Zosterella dubia***	***Filamentous algae***	UNKNOWN-1: [illegible]	UNKNOWN-2:
40	1	7587	4 0286		M						50		30														10													
	2				M			30																			50							20						
	3				M	20		10																			35							16						
38	1	7558	4 0402		M	25		20																			20							15				20		
	2				M	15		25					5														35							20						
	3				M	60		20					5														65													
33	1	7349	4 0952		M	2		10					T														88													
	2				M			25																										75						
	3				M	5		25			T																60							10						
29	1	6992	4 1293		S			[illegible]																			65							20						
	2				M	5		60			T		5														25							5						
	3				S			50																			40							10						
25	1	6942	4 1594		M	40		45																			8							7						
	2				M	10							3														7												80	
	3				M	10		70																										18			2			
21	1	6604	4 1728		Z																																			
	2				S			80			Z																							20						
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17	1	6360	4 1572		S			15			25		50														5							5						
	2				S			T			10																T							90						
	3				M			60			9		7																					30						
8	1	6460	4 0822		S	75		5																			[illegible]							20						
	2				M	60																					40													
	3				S	75		T			T																25													
7	1	6484	4 0768		M	99																					1													
	2				S	90																					10													
	3				M	88		T																			12													
2	1	6632	4 0336		M	5		50																			15							30						
	2				M	7		60																			20							13						
	3				S			40																			60													

Figure 5. Sampling form used to record point intercept rake toss relative abundance method (PIRTRAM) data.

Other materials were assembled to ensure consistent sampling and to facilitate storage of samples of unknown plant species (Hellquist, 1993). A sampling rake (Figure 6) comprised of the heads of two garden rakes wired together (13" long [33 cm]) was tied to 33' (10m) of woven nylon line (5/16 inch [0.8 cm] diameter; clothesline). An insulated container was cooled with ice and plastic zip lock bags and marking pens were packed. A large plastic tray (approximately 2′ x 2′ [0.6 m x 0.6m] like those used to collect condensation from refrigerator bottoms) was brought along for separating plant specimens by species.

Figure 6. Aquatic plant sampling rake comprised of the heads of two garden rakes wired together (13” long) and tied to 33’ of woven nylon line (5/16 inch diameter; clothesline).

With our arrival on the DeRuyter Reservoir, GPSs were activated and provided direct exposure to the equatorial (southern) sky, where GPS satellites are located. We proceeded to the first sample point as directed by the GPS and anchored within 15 feet (4.6 m) of the designated point and, with the end of the line attached to one hand, tossed the rake out from the boat approximately 30′ (9 m) towards the identified sampling point. We allowed the rake to fall to the bottom and, then, pulled the rake back to the boat at a pace that permitted rake contact with the bottom for as long as possible.

We evaluated the sample for overall abundance by association with a field measure as defined in Table 1 and noted the associated abundance category in the “All Plants Abundance” column of the sampling form (Figure 5). If the plant sample was “trace”, we recorded each plant species collected in the sample as “trace.” If it was not a trace sample, we took all plant material off the rake and placed it in the large plastic tray or on a clean deck area. Plant material was separated into piles by species until we could associate a percentage of the total plant material with each species. We recorded the percentage associated with any significant amount (> 1%) of plant material. If a species was present at less than ½%, we recorded it as “T.” If a species was unknown, we recorded it in one of the unknown columns and noted a description. We also stored a sample of each unknown plant on ice in a zip lock bag which had written on it the site, date, and collector. These were used to later identify the species. We tossed the rake a second and third time somewhat offset from the previous tosses and processed as noted above. We then proceeded to the next sample point and repeated the process.

Table 1. Abundance categories and their associated field measures and ranges of dry weights used in defining overall aquatic plant abundances when using the point intercept rake toss relative abundance plant method (PIRTRAM) for sampling.

Abundance Categories	Field Measure	Typical Dry Weight (g/m^2) Ranges associated with Plants Abundance
"Z" = no plant(s)	Nothing	0
"T" = trace plant(s)	Fingerful	~ 0.0001 - 2.000
"S" = sparse plant(s)	Handful	~ 2.001 - 140.000
"M" = medium plant(s)	Rakeful	~ 140.001 - 230.000
"D" = dense plant(s)	Can’t bring in boat	~ 230.001 - 450.000+

Accumulated data was keyed into a Cornell University Research Ponds authored Microsoft® Access® (2002) database created for mapping aquatic plant communities (*i.e.*, “DeRuyter Reservoir Rake Toss Samples as of ddmmmyy”). The database supports lake and day records and records for each sampling location as well as for each sample. When all data was entered, a query (*i.e*., “B1 All Plants Summary for ProLat & Delorme”) was run to produce extracts for all plants abundances. We then established a new Microsoft® Excel® (2013) spreadsheet (named the same as the database).

Translated data were copied back into the spreadsheet for DeRuyter Reservoir in separate worksheets for each abundance category: “D All Plants”, “M All Plants”, “S All Plants”, “T All Plants”, and “ZP All Plants.” Then, each worksheet was opened and data was deleted from each for any plant abundance records not matching the worksheet name, *e.g*., we deleted “D”,”M”, “S”, and “T” records from the “ZP All Plants” worksheet and deleted “M”, “S”, “T” and “ZP” records from the “D All Plants” worksheet. Results were saved.

Plant abundance symbols were created in a graphics program (Adobe Photoshop®) and were imported into our ESRI® ArcGIS Desktop (Version 9.3.1) and ArcMap™ (Version 9.3.1) custom ESRI symbol library. We then imported the data and associated an appropriate symbol with it and checked for mapped locations that might indicate a data entry error. Other data errors were identified by using a “Totals Query” which ensured percentages added up to 100% (or slightly greater if trace plant material was accounted for).

Maps produced by ArcMap were saved as files for use in this report.

Milfoil Stem Sample Collection and Processing

Milfoil apical meristems were collected to provide samples of the indigenous populations of milfoil insect herbivores and to ascertain what damage that those herbivores were causing milfoil in DeRuyter Reservoir. In 2018, we obtained 127 stems from various locations around the lake, on five different days, as noted in Figure 7 Milfoil stem samples were collected using the double headed plant rake previously described.   The rake was blindly tossed into a milfoil bed and extracted. One to two stems were collected off the rake with each toss. If more than two stems of sufficient length were on the rake, only the first two removed were used. To limit bias each stem was removed from the rake by locating the basal portion without viewing the apical tip. Samples were kept on ice until returned to the laboratory.

At the time of examination, we placed each stem sample under a stereoscopic dissecting microscope (Figure 8). We dissected each stem and evaluated the entire sample, recording numbers and types of herbivores found, evidence of herbivore use (retreats, cocoons, and pupa chambers), and milfoil tissue damage (leaflet damage, stem mining, missing or grazed apical meristems).

Due to the onset of a harmful algae bloom in mid-August, the basal milfoil samples were unable to be collected safely. As a diver could not be sent in safely to collect the samples we attempted to use a long rake to collect basal stems.

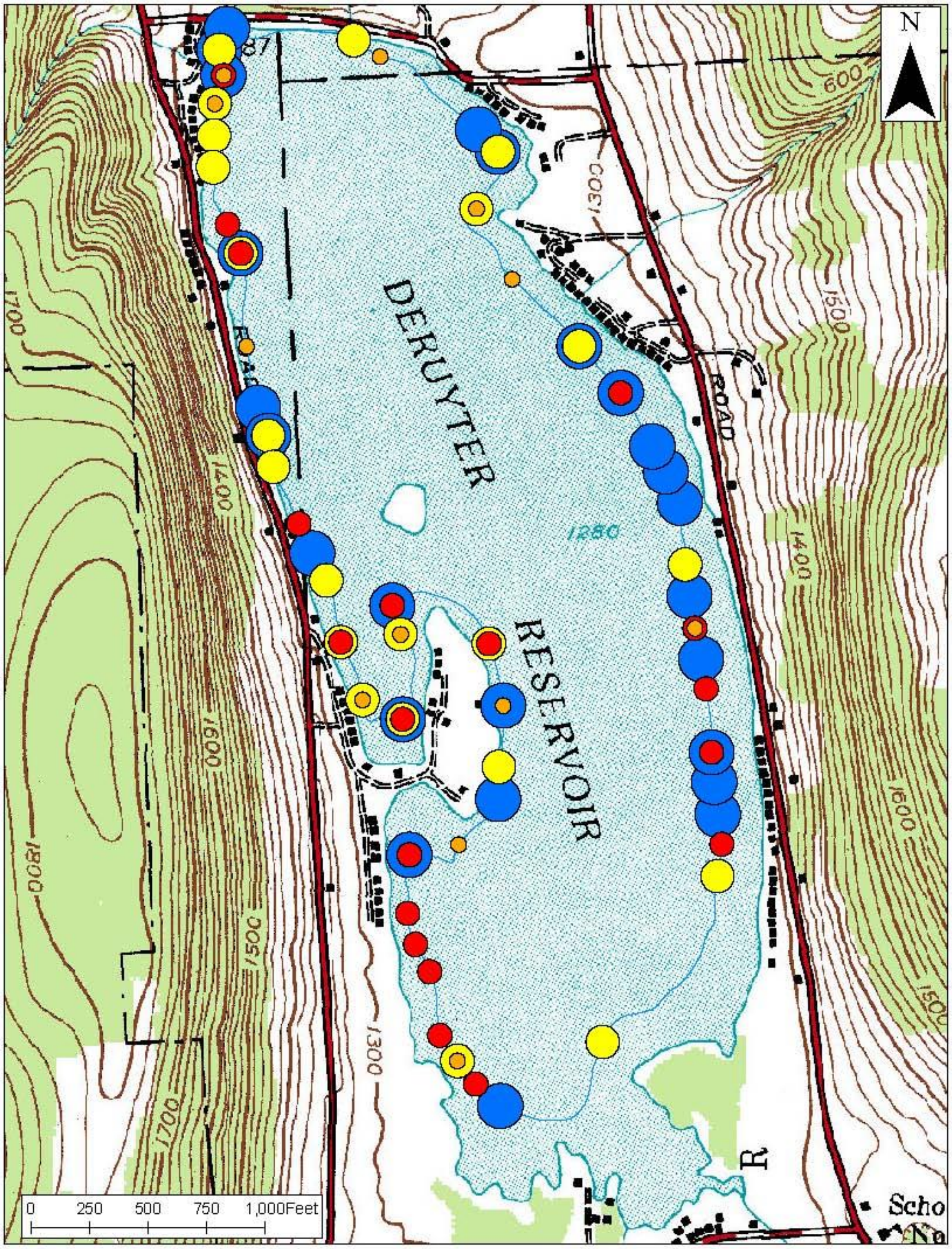

Figure 7. DeRuyter Reservoir collection sites used to evaluate herbivory and herbivore presence on stem tops of Eurasian watermilfoil (*Myriophyllum spicatum*) during 2018. All locations were originally randomly chosen from locations along the ten foot depth contour. Blue dots indicate June collection sites. Red dots indicate July collection sites. Yellow dots indicate August collection sites. Orange dots indicate September collection sites.

Figure 8. Dissecting microscopes used to observe herbivory and herbivore presence on stems of Eurasian watermilfoil (*Myriophyllum spicatum*).

Specific 2018 sampling sites are identified in Figure 7.

Electrofishing

SUNY Cobleskill completed electrofishing of DeRuyter Reservoir's warm water fish on 26 June 2018. This year a fall survey was performed on 25 October 2018. This additional survey was done to obtain more accurate estimates of gamefish populations. The methods used were in general conformance with NYSDEC, Division of Fish and Wildlife, Bureau of Fisheries guidelines (1989). The specific technique employed maneuvered the electrofishing boat parallel to the shoreline attempting to keep the boat in approximately 3 feet of water and moving into deeper water only when necessary to maneuver around boats, docks, and other obstructions. Specific sampling sites are identified in Figure 9. Specific June 2018 sampling sites are identified in Figure 9. 2 FIGURES NEEDED: ONE FOR JUNE & ONE FOR OCTOBER. They are the same sites used previously.

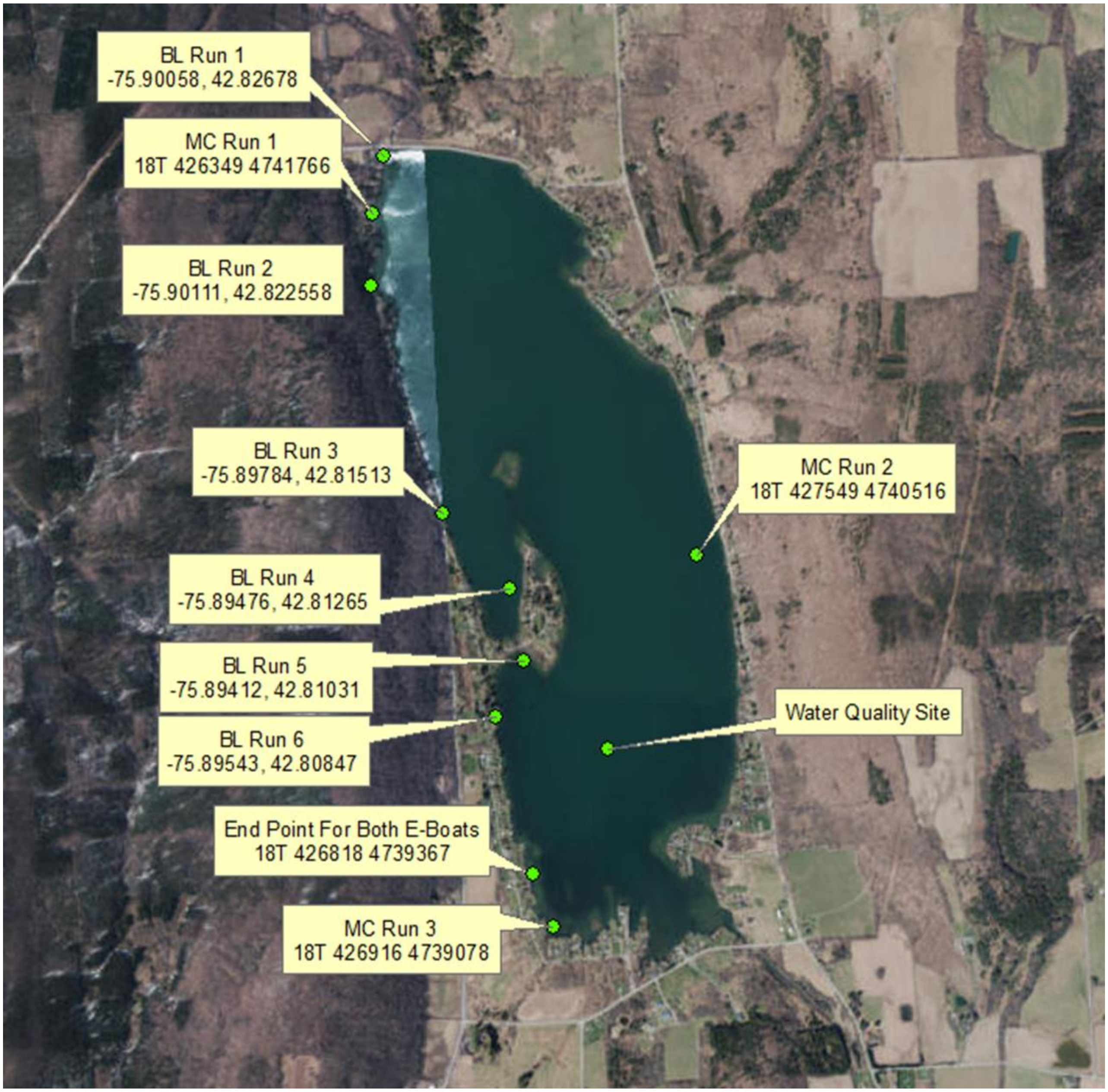

Figure 9. Locations of DeRuyter Reservoir electrofishing samples obtained on 26 June 2018.

Algae Monitoring

In August 2018, subsequent to observing a green sheen on the water's surface across parts of the lake (Figure 11), two water samples were collected in the northeast lake using a device that minimized human contact with the samples. The samples were provided to the New York State Department of Environmental Conservation's (NYSDEC) Harmful Algal Bloom (HAB) department for analysis (Figure 13).

Figure 10. Algal bloom observed at Snug Harbor, September 11, 2018.

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Results

Water Quality Sampling

Water quality data are displayed in Table 2. Water quality data from the October 17, 2018 sample are consistent with data from previous years and documents a consistent temperature from the surface to the bottom. Similar measurements, from the surface to the bottom, are evident in conductivity, pH and oxygen.

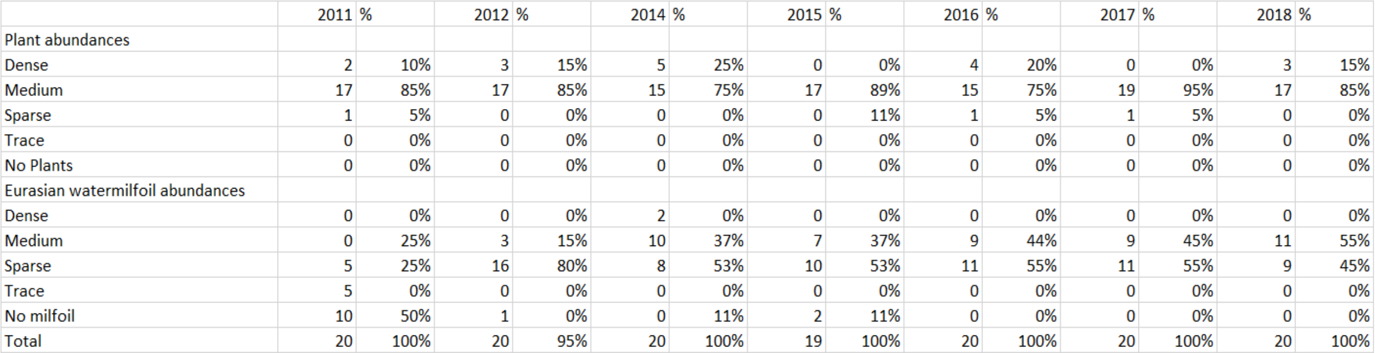
Table 2. Results of water quality samples obtained from site WQ2 on DeRuyter Reservoir as sampled on October 25, 2018 using a multi-probe device, calibrated per manufacturer's instructions, to measure water depth, temperature, conductivity, dissolved solids, salinity and oxygen and pH. m=meters; ^{o}C=Centigrade degrees; µS/cm=micro-Siemens per centimeter: g/L=grams per liter; ppt= parts per thousand; mg/L=milligrams per liter.

Water Quality						
	Temp^{o}C	DO (mg/L)	Cond (µS/cm)	TDS (mg/L)	Sal	pH
Surface	9.5	10.77	124.9	115.05	0.08	8.13
2	9.5	10.71	124.9	115.05	0.08	8.13
4	9.6	10.64	125.0	115.05	0.08	8.13
6	9.6	10.52	125.0	115.05	0.08	8.13
8	9.5	10.67	125.0	115.05	0.08	8.13
10	9.5	10.7	125.0	115.05	0.08	8.13
12	9.5	10.66	125.0	115.05	0.08	8.13
14	9.5	10.8	125.0	115.05	0.08	8.12
16	9.5	10.7	124.9	115.7	0.08	8.12
18	9.5	10.42	125.1	115.7	0.08	8.1
19	9.5	9.53	125.4	115.7	0.08	7.94

Macrophyte Sampling by the Point intercept rake toss relative abundance method (PIRTRAM)

Overall plant density in 2018 is slightly increased when compared to 2017 (Table 3). Eurasian milfoil density has changed minimally from 2017 to 2018 and was found at every site. The majority of Eurasian watermilfoil found was medium to sparse in density (100%). Point intercept rake toss relative abundance method (PIRTRAM) data for DeRuyter Reservoir as sampled in 2011, 2012, and 2014 - 2018 are summarized in Table 3. Figure 14 depicts the 2016 abundances of aquatic plants from PIRTRAM sampling while Figure 15 does the same for Eurasian watermilfoil. Appendix A provides detailed data from PIRTRAM sampling. Table 4 provides a summary of the aquatic plant species noted in DeRuyter Reservoir in all of our surveys & observations.

Table 3. Density summary for all plants and for Eurasian watermilfoil (*Myriophyllum spicatum*) in DeRuyter Reservoir as sampled from 2011, 2012, and 2014 through 2018 from the same randomly chosen locations along the 10-foot depth contour using the point intercept rake toss relative abundance method (PIRTRAM).



	2011	%	2012	%	2014	%	2015	%	2016	%	2017	%	2018	%
Plant abundances														
Dense	2	10%	3	15%	5	25%	0	0%	4	20%	0	0%	3	15%
Medium	17	85%	17	85%	15	75%	17	89%	15	75%	19	95%	17	85%
Sparse	1	5%	0	0%	0	0%	0	11%	1	5%	1	5%	0	0%
Trace	0	0%	0	0%	0	0%	0	0%	0	0%	0	0%	0	0%
No Plants	0	0%	0	0%	0	0%	0	0%	0	0%	0	0%	0	0%
Eurasian watermilfoil abundances														
Dense	0	0%	0	0%	2	0%	0	0%	0	0%	0	0%	0	0%
Medium	0	25%	3	15%	10	37%	7	37%	9	44%	9	45%	11	55%
Sparse	5	25%	16	80%	8	53%	10	53%	11	55%	11	55%	9	45%
Trace	5	0%	0	0%	0	0%	0	0%	0	0%	0	0%	0	0%
No milfoil	10	50%	1	0%	0	11%	2	11%	0	0%	0	0%	0	0%
Total	20	100%	20	95%	20	100%	19	100%	20	100%	20	100%	20	100%

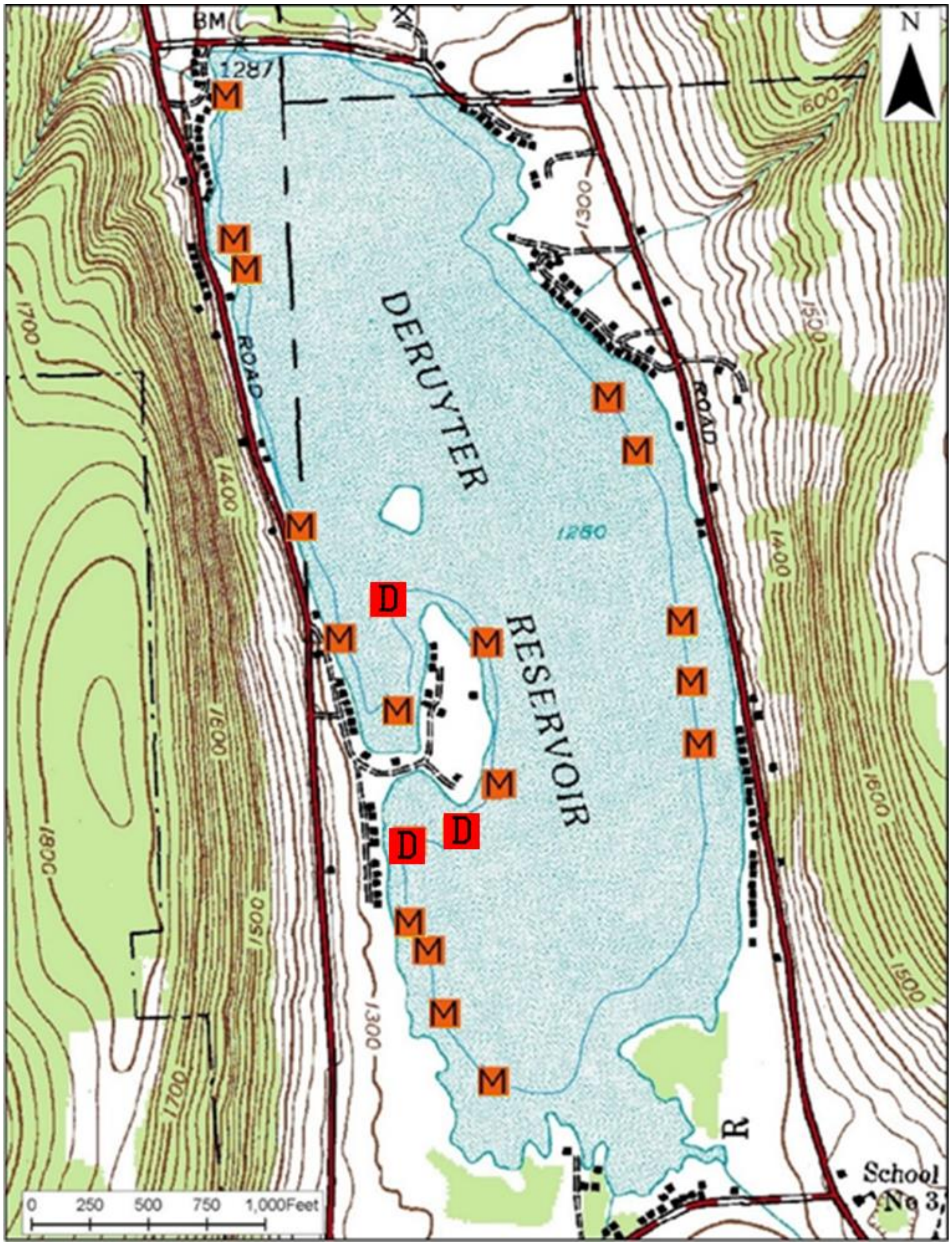


Figure 11. All aquatic plants abundances and locations in DeRuyter Reservoir from point intercept rake toss relative abundance method (PIRTRAM) samples obtained July 2018. "D" = dense plants; "M" = medium plants; "S" = sparse plants. See "Methods and Rationale" p. 17, Table 1 for weights associated with abundance categories and methods details.

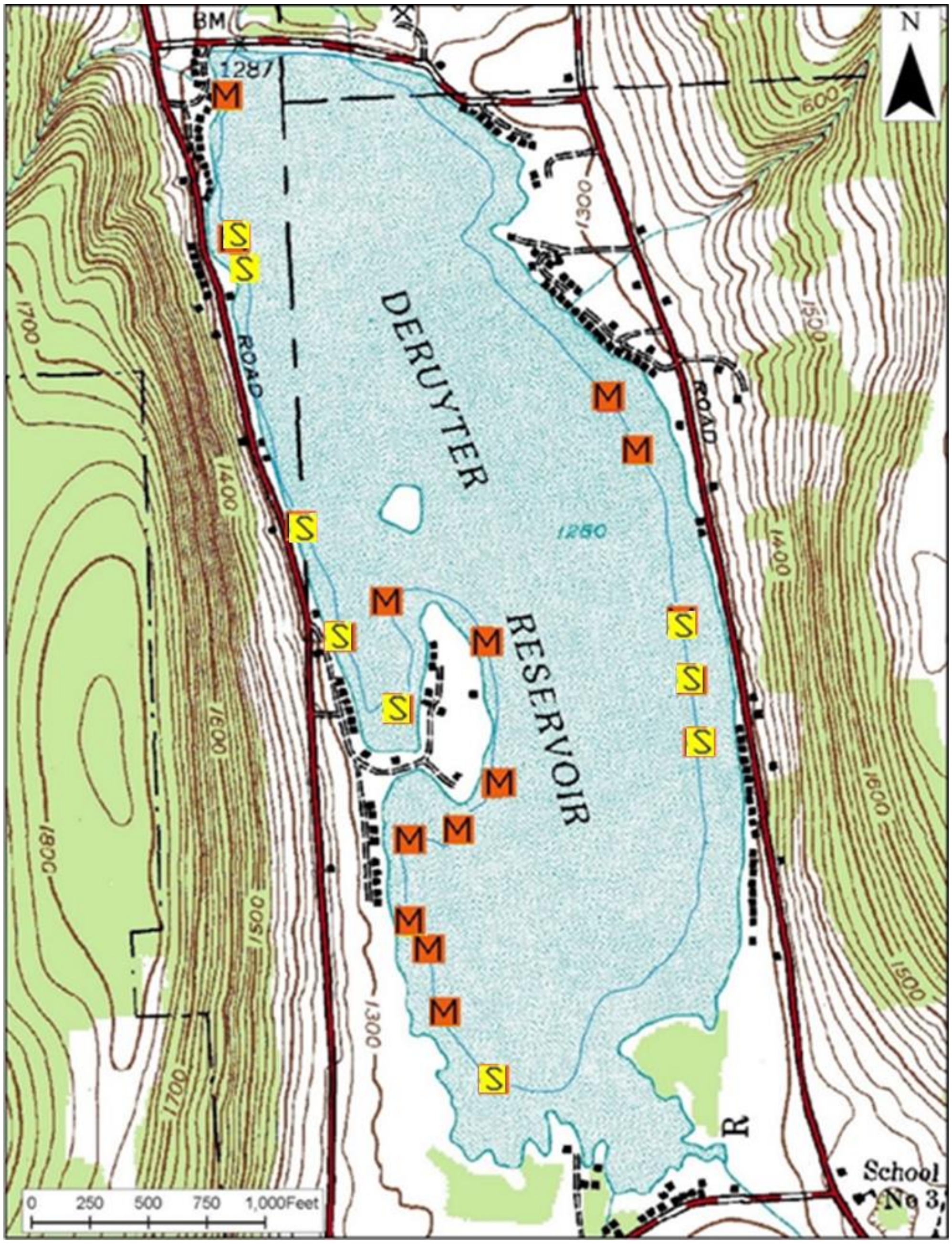


Figure 12. Eurasian watermilfoil (*Myriophyllum spicatum*) abundance and locations in DeRuyter Reservoir from point intercept rake toss relative abundance method (PIRTRAM) samples obtained July 2018. "S" = sparse Eurasian watermilfoil; "T"= trace Eurasian watermilfoil; "Circle Z" = no Eurasian watermilfoil. See "Methods and Rationale" p. 17, Table 1 for weights associated with abundance categories and methods details.

Table 4. Names of all submersed plants and the three macro algae found in DeRuyter Reservoir as sampled in 2011, 2012, and 2014 through 2018 from all samplings and observations.

	Common Name	Scientific Name
1	Clasping leaf pondweed	*Potamogeton richardsonii*
2	Coontail	*Ceratophyllum demersum*
3	Curly leaved pondweed	*Potamogeton crispus*
4	Eurasian watermilfoil	*Myriophyllum spicatum*
5	Filamentous algae	
6	Flat-stemmed pondweed	*Potamogeton zosteriformis*
7	Hybrid pondweed	*Potamogeton* sp. x sp.
8	Illinois pondweed	*Potamogeton illinoensis*
9	Nitella	*Nitella sp.*
10	Sago pondweed	*Stuckenia pectinata*
11	Small pondweed	*Potamogeton pusillus*
12	Slender naiad	*Najas flexilis*
13	Southern naiad	*Najas guadalupensis*
14	Starry Stonewort	*Nitellopsis obtusa*
15	Stonewort	*Chara vulgaris*
16	Tape-grass	*Vallisneria americana*
17	Water-crowfoot	*Ranunculus trichophyllus*
18	Water-plantain	*Alisma gramineum*
19	Water stargrass	*Heteranthera dubia*
20	Waterweed	*Elodea* sp.

Milfoil Stem Sample Collection and Processing

Milfoil herbivore data document increased numbers of milfoil herbivores in DeRuyter Reservoir in 2018 from 2017. Data from our milfoil stem sampling are summarized in Table 5. We found two aquatic macrophyte moth larvae, three moth eggs, one adult weevil, five weevil larvae, one weevil egg. We found milfoil midges (*Cricotopus myriophylli*) in all sampling periods (June = 24, 0.45 per stem; July = 36, 0.69 per stem; August = 15, 0.29 per stem; September = 43, 1.59 per stem). *Cricotopus myriophylli* numbers were greater by more than 0.5% from 2017, though numbers decreased in July and August.

Table 5. Results of Eurasian watermilfoil (*Myriophyllum spicatum*) stem samples obtained from DeRuyter Reservoir in June, July, August and October 2018. One hundred and twenty-eight stem samples were collected. Presence of aquatic macrophyte moths (*Acentria ephemerella*), milfoil midges (*Cricotopus myriophylli*), and milfoil weevils (*Euhrychiopsis lecontei*) in any life stage were noted.

Date	**Tip (#)**	**Moth Adult (#)**	**Moth LARVAE (#)**	**Pupae in cocoons (#)**	**Eggs (#)**	**Weevil Adult (#)**	**Weevil LARVAE (#)**	**Pupae in pupal chamber (#)**	**Eggs (#)**	**Midges non-damaging (#)**	**Midges DAMAGING Milfoil (#)**	**Caddisflies (#)**
29-Jun-18	27	0	1	0	0	0	1	0	0	187	24	29
# per tip		0.00	0.02	0.00	0.00	0.00	0.02	0.00	0.00	3.53	0.45	0.55
24-Jul-18	51	0	1	1	1	0	1	0	1	170	36	33
# per tip		0.00	0.02	0.02	0.02	0.00	0.02	0.00	0.02	3.27	0.69	0.63
20-Aug-18	24	0	0	0	0	0	1	0	0	410	15	15
# per tip		0.00	0.00	0.00	0.00	0.00	0.02	0.00	0.00	8.04	0.29	0.29
21-Sep-18	25	0	0	0	2	1	2	0	0	851	43	24
# per tip		0.00	0.00	0.00	0.07	0.04	0.07	0.00	0.00	31.52	1.59	0.89

Table 5 continued. Results of Eurasian watermilfoil (*Myriophyllum spicatum*) stem samples obtained from DeRuyter Reservoir in June, July, August and October 2018. One hundred and twenty-six stem samples were collected. Presence of aquatic macrophyte moths (*Acentria ephemerella*), milfoil midges (*Cricotopus myriophylli*), and milfoil weevils (*Euhrychiopsis lecontei*) in any life stage were noted.

Date	**Tip (#)**	**Moth Adult (#)**	**Moth LARVAE (#)**	**Pupae in cocoons (#)**	**Eggs (#)**	**Weevil Adult (#)**	**Weevil LARVAE (#)**	**Pupae in pupal chamber (#)**	**Eggs (#)**	**Midges non-damaging (#)**	**Midges DAMAGING Milfoil (#)**	**Caddisflies (#)**	**Healthy apical tip**	**Minor (a few leaflets missing)**	**Moderate (many leaflets gone)**	**Extensive (most grazed/deformed)**	**Tip Missing (end of stem)**	**Healthy Stem**	**Minor (1-4 scars or <cm mined)**	**Moderate (>4 scars or 2-4 cm mined)**	**Extensive (8 scars or >4 cm mined)**	**Weevil mining (#)**	**Pupal chambers (#)**	**Scars (# moth, weevil, or other)**	**Healthy Leaflets**	**Minor (leaflets gone in 1 place)**	**Moderate (leaflets gone, 2-4 places)**	**Extensive (leaflets gone, >4 places)**	**Retreats (# total)**	**Cocoons (#)**
29-Jun-18	27	0	1	0	0	0	1	0	0	187	24	29	0	5	4	29	3	13	12	2	0	6	0	16	0	3	0	24	50	0
# per tip		0.00	0.02	0.00	0.00	0.00	0.02	0.00	0.00	3.53	0.45	0.55	0.00	0.09	0.08	0.55	0.06	0.25	0.23	0.04	0.00	0.11	0.00	0.30	0.00	0.06	0.00	0.45	0.94	0.00
24-Jul-18	51	0	1	1	10	0	1	0	1	170	36	33	46	15	2	33	12	40	12	1	0	7	0	5	2	5	5	39	28	0
# per tip		0.00	0.02	0.02	0.19	0.00	0.02	0.00	0.02	3.27	0.69	0.63	0.88	0.29	0.04	0.63	0.23	0.77	0.23	0.02	0.00	0.13	0.00	0.10	0.04	0.10	0.10	0.75	0.54	0.00
20-Aug-18	24	0	0	0	0	0	1	0	0	410	15	15	25	2	0	14	7	18	6	0	0	3	1	4	1	6	5	12	35	0
# per tip		0.00	0.00	0.00	0.00	0.00	0.02	0.00	0.00	8.04	0.29	0.29	0.49	0.04	0.00	0.27	0.14	0.35	0.12	0.00	0.00	0.06	0.02	0.08	0.02	0.12	0.10	0.24	0.69	0.00
21-Sep-18	25	0	0	0	2	1	2	0	0	851	43	24	8	4	0	24	10	18	6	0	1	3	4	11	2	1	0	21	14	0
# per tip		0.00	0.00	0.00	0.07	0.04	0.07	0.00	0.00	31.52	1.59	0.89	0.30	0.15	0.00	0.89	0.37	0.67	0.22	0.00	0.04	0.11	0.15	0.41	0.07	0.04	0.00	0.78	0.52	0.00

Electrofishing.

The summer of 2018 had a lower percentage of sunfish (49%) compared to 2017. Table 6 shows the results of the summer and fall surveys in 2018. Bluegill total numbers decreased in the summer of 2018 when compared other summer surveys. Table 7 provides the electrofishing summaries for 2018 and preceding years, and Figure 16 displays graphs depicting our electrofishing sample of bluegill population composition by size for both 2018 and 2017. Catch per unit effort (catch per hour) for bluegill decreased from 249 fish per hour in 2017 to 63 fish per hour in summer of 2018. Pumpkinseed decreased from 194 per hour in 2017 to 67 per hour in the summer of 2018. Largemouth bass decreased from 74 fish per hour in 2017 to 19 fish per hour in the summer of 2018. The only distinct age class in both the summer and fall surveys is the young-of-the-year.

Table 6. Electrofishing data collected in the summer and fall of 2018.

Species	Catch/hr	Catch/hr
	6/26/2018	10/25/2018
Brown Bullhead	2.86	0
Black Crappie	0.29	2.67
Bluegill	63.14	0.00
Banded Killfish	0.00	0.00
Bluntnose Minnow	7.71	0.00
Common Carp	0.00	0.00
Chain Pickerel	2.86	32.33
Emerald Shiner	0.00	0.00
Golden Shine	6.00	0.00
Largemouth Bass	19.14	22.67
Central Mud Minnow	0.00	0.00
Pumpkin Seed	67.14	0.33
Rock Bass	53.71	0.33
Smallmouth Bass	2.00	1.33
Spottail Shiner	0.00	0.00
Tessellated Darter	1.71	0.00
Walleye	0.29	4.33
White Sucker	0.00	0.33
Yellow Perch	50.86	0
Yellow Bullhead	0.00	0.00
Total Fish	934.00	184
Total Sunfish	456.00	1
Sunfish as a % of total fish	0.49	0.01
Walleye as a % of total fish	0.00	0.07

Table 7. Electrofishing data summarized from seven years (2008, 2011, 2012 and 2014 through 2018) in DeRuyter Reservoir.

Species	Catch/hr	Catch/hr	Catch/hr	Catch/hr	Catch/hr	Catch/hr	Catch/hr	Catch/hr	7 Year	7 Year
	7/1/2008	6/30/2011	6/27/2012	6/26/2014	6/24/2015	7/11/2016	6/28/2017	6/26/2018	Average	Standard deviation
Brown Bullhead	4.7	7.4	1.1	11.4	10.7	6.6	6.3	2.86	6.38	2.64
Black Crappie	0	0	0	0.9	0.8	0	2.3	0.29	0.54	0.53
Bluegill	100.2	131	101.3	156.8	129.4	170.4	249.18	63.14	137.68	40.84
Banded Killfish	8.2	9.2	2.3	2.6	1.6	1.9	0	0.00	3.23	2.43
Bluntnose Minnow	1.2	14.8	21.4	10.6	6.6	13.1	0.79	7.71	9.53	5.45
Common Carp	23.3	0	0	0	0	0	0	0.00	2.91	4.53
Chain Pickerel	0	39.7	18	13.2	7.4	8.4	14.24	2.86	12.97	8.31
Emerald Shiner	17.5	0	1.1	0	0	0	0	0.00	2.33	3.37
Golden Shiner	24.5	40.6	9	7.9	23.1	17.8	32.43	6.00	20.17	9.99
Largemouth Bass	26.8	52.6	31.5	587.4	30.5	58.1	74.36	19.14	110.05	106.08
Central Mud Minnow	0	0	0	0.9	0	0	0	0.00	0.11	0.20
Pumpkin Seed	152.6	98.7	47.3	298.5	186.3	283.7	193.8	67.14	166.01	66.28
Rock Bass	125.8	179	283.5	229	108.8	87.1	103.63	53.71	146.32	63.14
Smallmouth Bass	19.8	10.2	4.5	7.9	4.1	8.4	11.87	2.00	8.60	3.57
Spottail Shiner	15.1	0.9	6.8	32.6	0	1.9	7.12	0.00	8.05	7.90
Tessellated darter	8.2	5.5	10.1	6.2	4.1	7.5	3.96	1.71	5.91	1.86
Walleye	8.2	3.7	4.5	2.6	0.8	1.9	1.58	0.29	2.95	1.89
White Sucker	0	0.9	3.4	2.6	0.8	0	0.79	0.00	1.06	0.86
Yellow Perch	349.4	121.8	398.3	374.3	204.4	228.5	55.37	50.86	222.87	114.76
Yellow Bullhead	0	0	0	0	0	0.9	0	0.00	0.11	0.18
Total fish	885.5	716	944.1	1745.4	719.4	896.2	757.72	934.00	949.79	198.90
total sunfish	252.8	229.7	148.6	455.3	315.7	454.1	442.98	456.00	344.40	95.73
Sunfish as a % of total fish	29%	32%	16%	26%	44%	51%	58%	49%	38%	12%

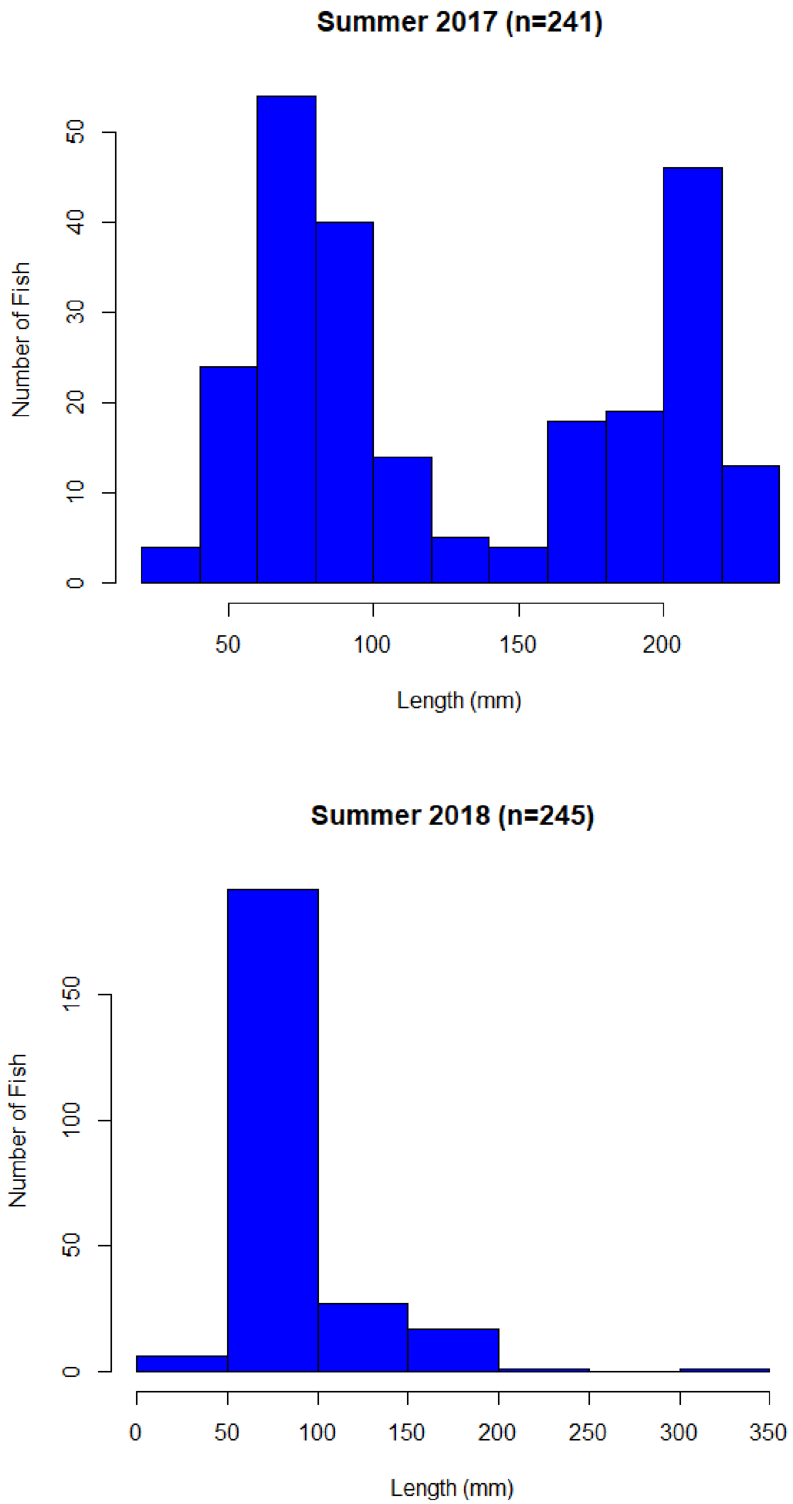


Figure 13. Length frequency histograms for bluegill (*Lepomis macrochirus*) in DeRuyter Reservoir for 2017 and 2018.

Algae monitoring

The taxa of algae and cyanobacteria identified by the NYSDEC, from water samples collected in August 2018 are shown in Table 8. The test showed levels of 60.26 µg/L (above the DEC Confirmed Bloom threshold of 25 µg/L) of *Dolichospermum* sp. (AKA *Anabaena* sp.) (NYSDEC Harmful Algae Bloom Program. 2018).

Table 8. Algae identified from DeRuyter Reservoir water samples collected on August 24, 2018.

Taxa	Description
Doliochospermum	Cyanobacteria formerly known as *Anabaena* sp.; often toxic.

Figure 14. *Doliocospermum* at 20X magnification. *Doliochospermum* is often toxic and are known to produce neurotoxins. (Picture by Kudela Lab, CA)

Discussion

Harmful algae blooms are increasing with frightening regularity in NY state. DeRuyter had seen it's first bloom in 2017 and another one this 2018. HABs not only limit recreation, but also interfere in our August sampling. Moving forward, we may need to consider alternative methods or an earlier sampling time to collect basal stems.

The decrease in panfish is encouraging, the lack of walleye, however, is concerning. Low stocking numbers and heavy fishing have likely impacted walleye numbers in DeRuyter Reservoir. 2018 the lowest observed number of walleyes in 10 years of stocking.

Recommendations

Fish Stocking

We again recommend an increase in walleye fingerlings stocked into DeRuyter Reservoir for 2019. Difficulties obtaining walleye in 2018 lead to low numbers being stocked this past December. Table 9 outlines the number of Walleye stocked in DeRuyter Reservoir for the last five years, and how it relates to the recommended stocking rates. We have been stocking DeRuyter Reservoir in the last three years at just about the NYS recommended stocking rate (when we average the fish stocked for each of those three years). Specifically, we recommend stocking with 45,000 walleye fingerlings in 2019 (Table 9) which includes our recommended stocking rate solution plus the accumulated deficiency from previous years' stockings. We recognize that finances may not permit walleye stocking at the recommended level and we are providing an estimated cost for stocking at 2/3 the recommended rate as well as for the recommended number. Please know that the data from less than recommended stockings are valuable to us. We hope that, for at least two years, DeRuyter Reservoir can be stocked at the recommended numbers so that we can create the change we all seek. After that, you should expect that you will be able to maintain control with about ½ the number that we are recommending for 2019.

If 2019 stocking should proceed with $^2/_3$ the recommended amount, that stocking should still produce modest improvement over what we saw in the last couple of years. With NYSDEC's recent increase in the minimum take length on DeRuyter Reservoir walleye to 18 inches and with a new maximum daily limit of three walleye (NYSDEC, undated), additional positive impacts regarding control of sunfish and increased milfoil herbivore populations should result.

Table 9. Comparison of Walleye fingerlings stocked in 2013 – 2018

Year	Recommended Stocking	Recommendation Relation to NYS Stocking Limit	Permit Authorized	Time of Stocking(s)	Number(s) Stocked	Stocking Relation to Recommended Stocking	Difference Between Numbers Stocked and NYS Stocking Limit	Actual Stocking Relation to NYS Stocking Limit
Goal		3x-6x						
2013	50600	4.5x	No Limit	Summer	50000	-600	38860	4.5x
2014	14000	1.3x	50000	Fall	10000	-4000	-41140	0.9x
2015	25000	2.2x	25000	Fall	25000	0	13860	2.2x
2016	36560	3.3x	25000	Fall	8000	-28560	-3140	0.7x
2017	37000	3.3x	36500	Fall	15000	-22000	3860	1.3x
2018	30000	2.7x	36500	Fall	18000	-12000	-18500	1.6x

Table 10. DeRuyter Reservoir 2018 walleye stocking considerations.

Walleye Finglerlings Calculation		Number	Notes
DeRuyter Reservoir acreage:		557	
NYS stocking rate:		20	/ acre
NYS stocking rate solution for DeRuyter Reservoir:		11,140	
Hypothetical insectivore controlling initial stocking rate:		5	x NYS Rate
	(not 10x because of previous stocking)		
	2013 summer fingerlings stocked:	50,000	(6,000 short of goal)
Hypothetical insectivore controlling maintenance stocking rate:		2	x NYS Rate with June fingerlings
	(adjusted by fish populations data)		
	2014 fall finglerlings stocked:	10,000	(~4,000 short of goal)
	2015 fall fingerlings stocked	25,000	
	2016 fall fingerlings stocked	8,000	(estimated not counted; ~14,000 short of goal)
	2017 fall fingerlings stocked	15,000	(estimated not counted; ~20,000 short of goal)
	2018 fall fingerlings stocked	18,000	(estimated not counted; ~12,000 short of goal)
DeRuyter 2019 fingerlings to be stocked:		45,000	(maintenance rate + 2018 deficit)
Fry required:		242,588	
2019 Oct walleye finglerlings estimated cost:		**$65,250**	($1.45/fingerling estimated)
Estimated cost for 30,000 October walleye fingerlings		**$43,500**	

Acknowledgements

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Appendices

Appendix A: Submersed aquatic plant relative abundances for DeRuyter Reservoir as sampled July 2018

Station	Zone	UTM NAD27 Northing	UTM NAD27 Easting	Depth (m)	All Plants	All Natives	Milfoil	Coontail	Stonewort	Yellow water lily	White water lily	Curly leaved pondweed	Leafy pondweed	Fern leaved pondweed	Water Stargrass
7	18T	4661511	0646416	3.0	M	M	S	S	Z	Z	Z	Z	Z	Z	Z
8	18T	4661442	0646447	3.0	M	M	S	M	Z	Z	Z	Z	Z	Z	Z
17	18T	4660794	0646586	3.0	M	M	S	M	Z	Z	Z	Z	Z	Z	Z
21	18T	4660510	0646687	3.0	M	M	S	M	Z	Z	Z	Z	Z	Z	T
25	18T	4660325	0646835	3.0	M	M	S	S	Z	Z	Z	S	Z	Z	S
29	18T	4660598	0646810	3.0	D	M	M	M	Z	Z	Z	Z	Z	Z	Z
33	18T	4660506	0647042	3.0	M	M	M	S	Z	Z	Z	Z	Z	Z	Z
38	18T	4660134	0647064	3.0	M	M	M	S	Z	Z	Z	Z	Z	Z	T
40	18T	4660025	0646969	3.0	D	M	M	M	Z	Z	Z	Z	Z	Z	S
42	18T	4660001	0646851	3.0	D	M	M	S	Z	Z	Z	Z	Z	Z	S
45	18T	4669786	0646866	3.0	M	S	M	S	Z	Z	Z	Z	Z	Z	T
46	18T	4669721	0646900	3.0	M	M	M	S	Z	Z	Z	Z	Z	Z	T
48	18T	4669568	0646924	3.0	M	M	M	S	Z	Z	Z	Z	Z	Z	T
51	18T	4669399	0647069	3.0	M	M	S	M	Z	Z	Z	Z	Z	Z	T
67	18T	4660246	0647575	3.0	M	M	S	S	Z	Z	Z	T	Z	Z	S
69	18T	4660398	0647561	3.0	M	M	S	S	Z	Z	Z	T	Z	Z	S
71	18T	4660544	0647535	3.0	M	M	S	M	Z	Z	Z	Z	Z	Z	T
77	18T	4660979	0647432	3.0	M	M	M	S	Z	Z	Z	S	Z	Z	S
79	18T	4664110	0642735	3.0	M	S	M	S	Z	Z	Z	Z	Z	Z	Z

Appendix B: Glossary

25 cm: 10 inches

ab·scis·sion

Pronunciation: ab-'si-zh&n
Function: *noun*
Etymology: Latin *abscission-, abscissio,* from *abscindere*
: the act or process of cutting off : REMOVAL;
: the natural separation of flowers, fruit, or leaves from plants at a special separation layer.

aer·en·chy·ma

Pronunciation: "ar-'e[ng]-k&-m&, "er-

Function: *noun*

Etymology: New Latin

: the spongy modified cork tissue of many aquatic plants that facilitates gaseous exchange and maintains buoyancy.

al·ga

Pronunciation: 'al-g&
Function: *noun*
Inflected Form(s): *plural* al·gae
Etymology: Latin, seaweed
: a plant or plantlike organism of any of several phyla, divisions, or classes of chiefly aquatic usually chlorophyll-containing nonvascular organisms.

api·cal

Pronunciation: 'A-pi-k&l *also* 'a-pi-
Function: *adjective*
Etymology: probably from New Latin *apicalis,* from Latin *apic-, apex*
: of, relating to, or situated at an apex.

aug·men·ta·tion
Pronunciation: "og-m&n-'tA-sh&n, -"men-
Function: *noun*
: the act or process of augmenting;

: the state of being augmented;
: something that augments.

aux·in

Pronunciation: 'ok-s&n
Function: *noun*
Etymology: from Greek *auxein*
: any of various usually acidic organic substances that promote cell. elongation in plant shoots and usually regulate other growth processes (as root initiation)

bas·al

Pronunciation: 'bA-s&l, -z&l
Function: *adjective*
: relating to, situated at, or forming the base

: arising from the base of a stem, e.g., *basal* leaves.

buoy·an·cy
Pronunciation: 'boi-&n(t)-sE, 'bü-y&n(t)-
Function: *noun*
: the tendency of a body to float or to rise when submerged in a fluid

: the power of a fluid to exert an upward force on a body placed in it; *also* : the upward force exerted.

can·o·py

Pronunciation: 'ka-n&-pE
Function: *noun*
Inflected Form(s): *plural* -pies
Etymology: Middle English *canope,* from Medieval Latin *canopeum* mosquito net, from Latin *conopeum,* from Greek *kOnOpion,* from *kOnOps*
: a protective covering: as

(1) **:** the uppermost spreading branchy layer of a forest or

(2) **:** parts of aquatic macrophytes floating horizontally at or near the water's surface.

ca·rot·en·oid
Variant(s): *also* ca·rot·in·oid /k&-'rä-t$^{\&}$n-"oid/
Function: *noun*
: any of various usually yellow to red pigments (as carotenes) found widely in plants and animals and characterized chemically by a long aliphatic polyene chain composed of eight isoprene units.
- carotenoid *adjective*.

dis·sect·ed vein
Pronunciation: di-'sekt-d; ÷dI-'sekt-d, ÷'dI-" 'vAn
Function: *noun*
Etymology: Latin *dissectus,* past participle of *dissecare* to cut apart, from *dis-* + *secare* to cut; Middle English *veine*, from Old French, from Latin *vena*
: vascular bundles forming the framework of a leaf with little or no leaf material between them.

eu·tro·phic
Pronunciation: yu-'trO-fik
Function: *adjective*
Etymology: probably from German *Eutroph* eutrophic, from Greek *eutrophos* well-nourished, nourishing, from *eu-* + *trephein* to nourish
of a body of water : characterized by the state resulting from eutrophication – compare to MESOTROPHIC.

ex·ot·ic
Pronunciation: ig-'zä-tik
Function: *adjective*
Etymology: Latin *exoticus,* from Greek *exOtikos,* from *exO*
: introduced from another country **:** not native to the place where found

ex·trap·o·late
Pronunciation: ik-'stra-p&-"lAt
Function: *verb*
Inflected Form(s): -lat·ed; -lat·ing
Etymology: Latin *extra* outside + English *-polate* (as in *interpolate*)
transitive senses

: to infer (values of a variable in an unobserved interval) from values within an already observed interval

- ex·trap·o·la·tion /-"stra-p&-'lA-sh&n/ *noun*.

fish·ery
Pronunciation: 'fi-sh&-rE
Function: *noun*
Inflected Form(s): *plural* -er·ies
: the occupation, recreation, industry, or season of taking fish or other sea animals (as sponges, shrimp, or seals) : FISHING.

ge·nus
Pronunciation: 'jE-n&s, 'je-
Function: *noun*
Inflected Form(s): *plural* gen·era /'je-n&-r&/
Etymology: Latin *gener-*, *genus* birth, race, kind
: a LWLAss, kind, or group marked by common characteristics or by one common characteristic; *specifically*

: a category of biological LWLAssification ranking between the family and the species, comprising structurally or phylogenetically related species or an isolated species exhibiting unusual differentiation, and being designated by a Latin or latinized capitalized singular noun.

her·bi·vore

Pronunciation: '(h)&r-b&-"vOr, -"vor
Function: *noun*
Etymology: New Latin *Herbivora*, group of mammals, from neuter plural of *herbivorus*
: a plant-eating animal.

her·biv·o·rous

Pronunciation: "(h)&r-'biv-r&s, -'bi-v&-
Function: *adjective*
Etymology: New Latin *herbivorus*, from Latin *herba* grass + *-vorus* -vorous
: feeding on plants.
- her·biv·o·ry /-'bi-v&-rE/ *noun.*

hy·poth·e·sis
Pronunciation: hI-'pä-th&-s&s
Function: *noun*
Inflected Form(s): *plural* hy·poth·e·ses
Etymology: Greek, from *hypotithenai* to put under, suppose, from *hypo-* + *tithenai* to put
: a tentative assumption made in order to draw out and test its logical or empirical consequences.

in·star
Pronunciation: 'in-"stär
Function: *noun*
Etymology: New Latin, from Latin, equivalent
: a stage in the life of an arthropod (such as an insect) between two successive molts; *also* **:** an individual in a specified instar.

lim·it·ing
Function: *adjective*
: functioning as a limit : RESTRICTIVE, e.g., *limiting* value;

: being an environmental factor (as a nutrient) that limits the population size of an organism.

lit·to·ral

Pronunciation: 'li-t&-r&l; "li-t&-'ral, -'räl
Function: *adjective*
Etymology: Latin *litoralis,* from *litor-*, *litus* seashore
: of, relating to, or situated or growing on or near a shore.

: that area of a lake or pond where the bottom is covered with macrophytes.

log·a·rithm
Pronunciation: 'lo-g&-"ri-[th]&m

Function: *noun*
Etymology: New Latin *logarithmus,* from *log-* + Greek *arithmos* number
: the exponent that indicates the power to which a number is raised to produce a given number, e.g., the *logarithm* of 100 to the base 10 is 2.

mac·ro·phyte

Pronunciation: 'ma-kr&-"fIt
Function: *noun*
: a member of the macroscopic plant life, especially of a body of water, i.e., plants growing in water that can be seen with the naked eye.

mer·i·stem
Pronunciation: 'mer-&-"stem
Function: *noun*
Etymology: Greek *meristos* divided (from *merizein* to divide, from *meros*) + English *-em* (as in *system*)
: a formative plant tissue usually made up of small cells capable of dividing indefinitely and giving rise to similar cells or to cells that differentiate to produce the definitive tissues and organs.

me·so·tro·phic
Pronunciation: "me-z&-'trO-fik, "mE-, -s&-, -'trä-fik
Function: *adjective*
of a body of water : having a moderate amount of dissolved nutrients -- compare EUTROPHIC.

mi·cro·bi·al
Pronunciation: /mI-'krO-bE-&l/
Function: *adjective*
Etymology: International Scientific Vocabulary *micr-* + Greek *bios* life
: pertaining to microorganism, germ.

midge
Pronunciation: 'mij
Function: *noun*
Etymology: Middle English *migge,* from Old English *mycg;* akin to Old High German *mucka* midge, Greek *myia* fly, Latin *musca*
Date: before 12th century
: a tiny dipteran fly (as a chironomid).

mu·tu·al·ism

Pronunciation: 'myü-ch&-w&-"li-z&m, 'myü-ch&-"li-, 'myüch-w&-"li-
Function: *noun*
: mutually beneficial association between different kinds of organisms

- mu·tu·al·is·tic /"myü-ch&-w&-'lis-tik, "myü-ch&-'lis-, "myüch-w&-'lis-/ *adjective*.

na·scent
Pronunciation: 'na-s$^{\&}$nt, 'nA-
Function: *adjective*
Etymology: Latin *nascent-*, *nascens*, present participle of *nasci* to be born
: coming or having recently come into existence.

niche

Pronunciation: 'nich, ÷'nEsh
Function: *noun*
Etymology: French, from Middle French, from nicher to nest, from (assumed) Vulgar Latin nidicare, from Latin nidus nest -- more at NEST
: a habitat supplying the factors necessary for the existence of an organism or species

: the ecological role of an organism in a community especially in regard to food consumption.

over·win·ter
Pronunciation: "O-v&r-'win-t&r
Function: *intransitive verb*
: to survive the winter.

plank·ton
Pronunciation: 'pla[ng](k)-t&n, -"tän
Function: *noun*

Etymology: German, from Greek, neuter of *planktos* drifting, from *plazesthai* to wander, drift, middle voice of *plazein* to drive astray; akin to Latin *plangere* to strike
: the passively floating or weakly swimming usually minute animal and plant life of a body of water
- plank·ton·ic /pla[ng](k)-'tä-nik/ adjective.

pre·da·tion

Pronunciation: pri-'dA-sh&n

Function: *noun*

Etymology: Middle English *predacion*, from Latin *praedation-*, *praedatio*, from *praedari*

: a mode of life in which food is primarily obtained by the killing and consuming of animals.

quad·rat
Pronunciation: 'kwä-dr&t, -"drat
Function: *noun*
Etymology: alteration of quadrate
: a usually rectangular plot used for ecological or population studies.

ref·uge
Pronunciation: 're-(")fyüj *also* -(")fyüzh
Function: *noun*
Etymology: Middle English, from Middle French, from Latin *refugium,* from *refugere* to escape, from *re-* + *fugere* to flee
: shelter or protection from danger or distress
: a place that provides shelter or protection.

rhi·zome
Pronunciation: 'rI-"zOm
Function: *noun*
Etymology: New Latin *rhizomat-, rhizoma,* from Greek *rhizOmat-, rhizOma* mass of roots, from *rhizoun* to cause to take root, from *rhiza* root

: a somewhat elongate usually horizontal subterranean plant stem that is often thickened by deposits of reserve food material, produces shoots above and roots below, and is distinguished from a true root in possessing buds, nodes, and usually scalelike leaves.

Sec·chi disk

Pronunciation: se-'kee desk
Function: *noun*
: disk used to measure water LWLArity.

se·nes·cence
Pronunciation: si-'ne-s$^{\&}$n(t)s
Function: *noun*

Etymology: *senescent*, from Latin *senescent-*, *senescens*, present participle of *senescere* to grow old, from *sen-*, *senex* old
: the state of being old : the process of becoming old.

: the growth phase in a plant or plant part (as a leaf) from full maturity to death
- se·nes·cent /-s&nt/ *adjective.*

sub·strate
Pronunciation: 's&b-"strAt
Function: *noun*
Etymology: Medieval Latin *substratum*
: the base on which an organism lives, e.g., the soil is the *substrate* of most seed plants.

tur·i·on

Pronunciation: ter-'E-on

Function: *noun*

: a winter bud arising from vegetative material.

un·sus·tain·able
Pronunciation: n-s&s-'stA-n&-b&l
Function: *adjective*
: incapable of being sustained.

: of or relating to a lifestyle involving the use of sustainable methods, e.g., *unsustainable* society.

Most of the definitions in this appendix were modified from Merriam-Webster OnLine: The Language Center (http://www.m-w.com.)