

Note

A new device for sampling submersed aquatic plants using underwater video

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INTRODUCTION

Measurement and examination of submersed aquatic plants (e.g., their abundance, growth, and morphology) is challenging because approaches typically applied in terrestrial systems are not easily transferrable to the aquatic environment. For example, quadrat-based methods conducted underwater typically require expensive scuba equipment and trained divers (Johnson and Newman 2011). The difficulty in sampling submersed vegetation has led to the development of a variety of methods to sample plants below the water's surface, and these methods generally fall into two broad categories: destructive and nondestructive (Madsen and Wersal 2017). Destructive sampling typically involves the removal of plant material via rakes, corers, dredges, or box samplers (Crowell et al. 1994, Madsen et al. 2007, Johnson and Newman 2011, Madsen and Wersal 2017); removed plants are then identified and weighed. Nondestructive sampling involves measuring submersed plants *in situ* through visual observations while snorkeling or diving, or by use of hydroacoustic technologies (Sheldon and Boylen 1978, Maceina et al. 1984, Madsen and Wersal 2017). Despite their importance for aquatic plant sampling, common destructive and nondestructive sampling methods are not ideal for all applications.

Although destructive sampling methods allow for simple, rapid, and repeatable measurement of submersed plants, they have drawbacks for some situations. For example, studies assessing efficacy of management typically include estimation of target species abundance over time (e.g., before and after treatment; Johnson et al. 2012, Parks et al. 2016, Glisson et al. 2018). Studies assessing seasonal growth and phenology also include estimation of abundance over time (e.g., Madsen 1997, Wersal et al. 2011, Marko et al. 2015). Continual removal of plants from the same locations over the course of such studies may result in biased estimates of abundance; thus, destructive sampling is not recommended for such applications (Madsen 1993). Repeated destructive sampling may also negatively impact nontar-

get native plants, including rare or threatened species (Madsen and Wersal 2017). Destructive sampling can also increase the risk of invasive species spread within and among lakes by producing fragments that can drift to new areas or be subject to overland transport (Rothlisberger et al. 2010). Lastly, some metrics, such as submersed plant height and morphology (e.g., branching and growth form), are not measureable with typical destructive sampling methods.

Some of these issues can be overcome with nondestructive sampling methods, but these methods can introduce other tradeoffs that limit their applicability. For example, scuba-based sampling of submersed plants (Parsons et al. 2009, Brainard and Schulz 2016, Thum et al. 2017) requires highly trained personnel, expensive equipment, and specific conditions for safety and efficacy (e.g., temperature, clarity, and vegetation density). Where scuba diving is not feasible, nondestructive sampling from the surface can be performed using hydroacoustic technology to estimate height, distribution, and biovolume of submersed vegetation (Madsen and Wersal 2017). This approach is useful for estimating total submersed plant growth and is highly efficient for coverage of large areas (Valley et al. 2005). However, hydroacoustic methods cannot be used to differentiate species (Maceina et al. 1984, Sabol et al. 2009, Valley et al. 2015) and are thus not well suited for detecting changes in individual target species or community composition. Additionally, in temperate environments, hydroacoustics and scuba diving are primarily limited to warm-season sampling due to ice cover.

To address some of the limitations of commonly used destructive and nondestructive methods and enrich the submersed plant sampling tool kit, we developed a novel sampling device that efficiently incorporates underwater video to measure species-specific abundance, growth, height, and morphology of submersed plants that can be used year round (e.g., for under-ice phenological studies). Our device provides the visual acuity of underwater observation, while allowing the observer to remain above the water's surface. It can be operated from any stable platform (e.g., boat, dock, or ice), is simple to use, and has wide applicability. We explain the design and use of this sampling device, illustrate its utility with data collected from field observations of watermilfoil (*Myriophyllum* spp.),

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and suggest other sampling situations to which it could be applied.

MATERIALS AND METHODS

Design and construction

The sampling device consists of an aluminum metal sampling frame that can move up and down sets of two aluminum poles¹ via rubber wheels (Figures 1 and 2). Each set of aluminum poles is bolted together and has button clips at the bottom to allow for multiple sets of poles to be combined to extend their length (Figure 2A). The poles are marked at 0.5-m intervals with yellow electrical tape and include a 10 by 20-cm rubber base to prevent the poles from sinking into the substrate (Figure 2). A rope is attached at the top of the metal frame to lower and lift the frame into and out of the water (Figure 2C). The rope is also marked at 0.5-m intervals that can be lined up with the marks on the poles. The metal frame has a small platform for a waterproof camera that faces a white plastic board attached to the frame by cable ties (Figure 2D). The plastic board has a 30 by 70-cm rectangle marked by black electrical tape in the center, which comprises the sampling window (Figure 2B). Wire mesh is attached with cable ties to the metal frame around the camera platform to reduce obstruction of the camera by vegetation. A waterproof camera is affixed to the camera platform and linked via a cable to a tablet computer that remains above the surface (i.e., on a boat, dock, or ice) as the sampling device is submerged. We used a GoPro camera² and an iPad tablet³ and the software associated with these devices; however, other waterproof cameras and tablets could also be used. The underwater camera must be Wi-Fi™ and Bluetooth® capable in order to transmit the video feed to the tablet computer in real time. Other underwater camera systems, such as those developed for ice fishing, could be used with the device; however, these may lack the versatility and functionality of the separate GoPro and iPad components. Unlike some camera systems developed for ice fishing, the GoPro camera and iPad tablet are not permanently attached and thus can be used separately for ongoing or future work in and out of the water. Additionally, cameras developed for ice fishing may not have the ability to record video, offer associated video-editing software, or have the functionality of GoPro software.

As Wi-Fi™ cannot transmit through water more than several centimeters, we used a multi-strand coaxial cable (RG 135) to carry the wireless signal through the water from the camera to the tablet computer (Figures 2 and 3). To accomplish this, we removed ~ 10 cm of the multi-strand coaxial cable's plastic jacket and conductive shield at each end (keeping the inner wire and its tubular insulator intact). With the ends of the cable unshielded, the cable functions as an antenna to transmit the Wi-Fi™ signal from the underwater camera to the tablet computer above the water. We created a watertight seal around the exposed section of the cable by placing a small piece of heat-shrink tubing where the plastic jacket and conductive shielding were cut. We then placed the exposed sections between the adhesive

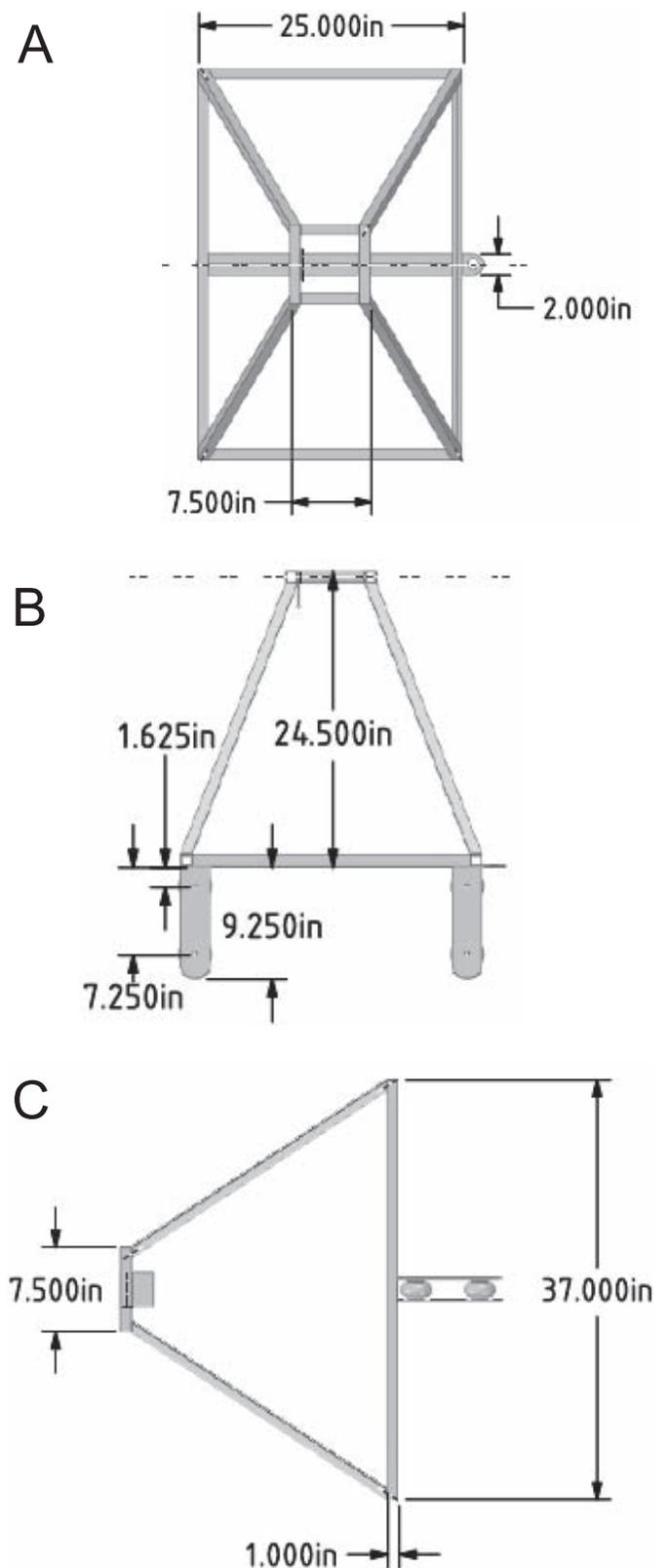


Figure 1. Technical drawing of metal sampling frame used in the sampling device: (A) front view, (B) side view, and (C) top view. The sampling frame is constructed from 2.5-cm square aluminum tubing with 3.175-mm walls.

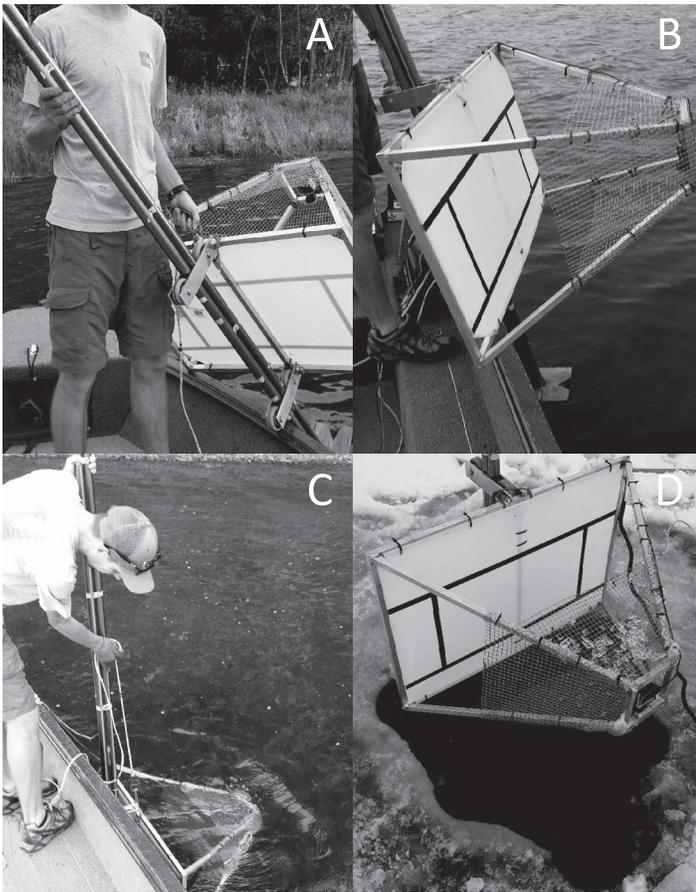


Figure 2. Images of the design and use of the sampling device, including (A, B, and C) summer and (D) winter use.

sides of two pieces of heavy-duty, Velcro®-like fasteners,⁴ which were used to adhere the exposed ends to the camera and tablet computer. Fasteners were then affixed to waterproof cases on both the camera and tablet, such that the cable could be easily attached to both devices (Figure 3). Once connected with the cable, the camera can be operated via software on the tablet computer, regardless of water depth. The total cost of all the components used to construct and employ the sampling device, including the camera and tablet, was approximately \$1,500. The electronics and associated waterproof cases comprised the majority of this cost (ca. \$1,200) and the mechanical components (i.e., materials for frame construction, poles, and cable setup) comprised the remainder. Construction required welding the aluminum frame together, and drilling holes in the plastic board (to insert the cable ties) and the aluminum poles (to bolt them together).

Field implementation

The sampling device could potentially be used for a variety of applications but was initially designed to monitor growth, phenology, and morphology of northern, Eurasian, and hybrid watermilfoils (*Myriophyllum sibiricum* Kon., *Myriophyllum spicatum* L., and *M. spicatum* × *M. sibiricum*; hereafter, watermilfoil) in Minnesota lakes. To demonstrate

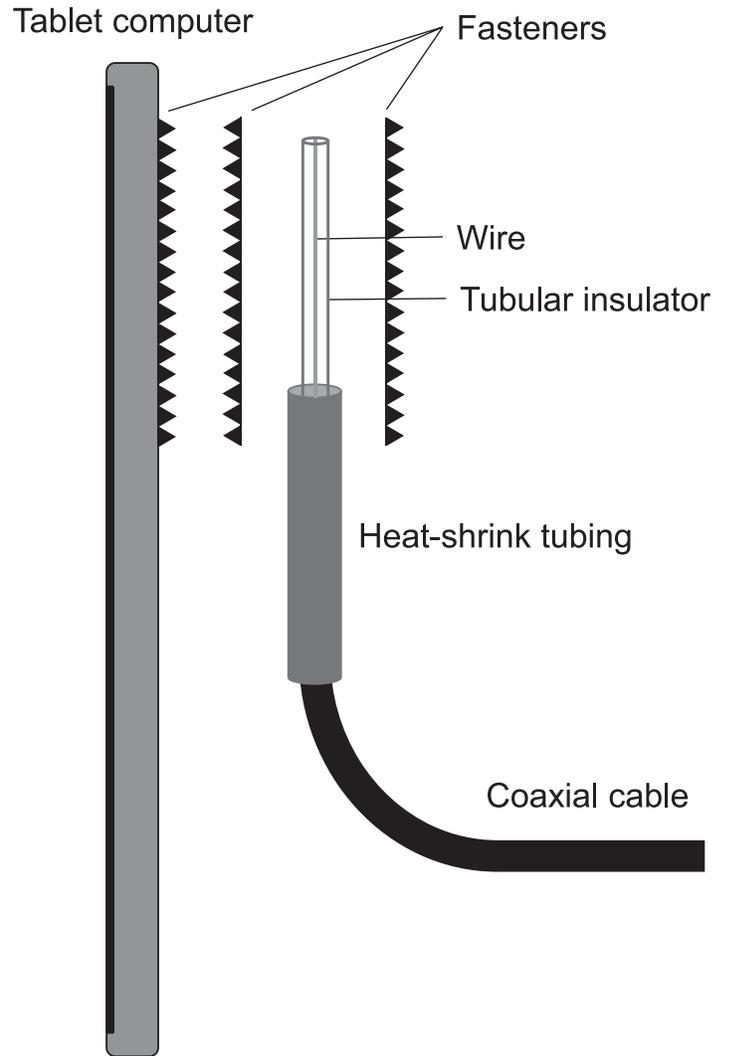


Figure 3. Construction and attachment of coaxial cable to tablet computer used with the sampling device. The coaxial cable is used to transmit a Wi-Fi™ signal from the underwater camera to the tablet computer above the water so that the underwater camera can be controlled and the video feed viewed and annotated in real time.

application of our sampling device, we describe the general methods used in this study for illustrative purposes.

We selected five points within watermilfoil beds (hereafter, sites) on five lakes in the Twin Cities, Minnesota, metro area (Big Carnelian Lake, Washington County; Cedar Lake, Hennepin County; Otter Lake, Anoka County; Orchard Lake, Dakota County; and Lake Phalen, Ramsey County). We visited these lakes every 2 to 4 wk during the growing season (June to November) in 2017 and once in winter (January or February) in 2018. We visited each lake a total of eight or nine times throughout this period.

Upon arrival at each site, we set two or more anchors to maintain our position and minimize boat movement. We then collected four separate video subsamples with the sampling device off the sides of the boat to account for spatial heterogeneity in watermilfoil presence and abundance. Subsamples were positioned at fore and aft port and fore and aft starboard, and separated by 2 to 3 m. To deploy

the sampling device, we held the device over the side of the boat and let the poles slide through the frame and onto the substrate (Figure 2B). We then turned on the GoPro camera and started recording video using the GoPro mobile application on the tablet computer. Once the camera was recording, we identified each video with its subsample number (1 to 4) by placing a hand in front of the camera and holding up the corresponding number of fingers. We then held the metal frame at the first height marker above the water's surface by matching marks on the rope with the labeled marks on the poles (Figure 2C). We recorded this initial height on a data sheet and placed a digital mark in the video file using the "Highlight" function on the GoPro mobile application; this places a yellow indicator mark on the video that can be seen on the GoPro mobile application or when the video is displayed in GoPro Studio software on a desktop computer. We then slowly lowered the frame into the water, stopping at 0.5-m intervals along the poles and marking the video with the Highlight function at each 0.5-m interval until we reached the substrate, which we denoted with two consecutive Highlight marks. We note that, because our sampling window was centered on the plastic board to ensure visibility, the minimum height at which plants could be observed was 15 cm above the substrate. Also, if plants were moving in the sampling window at a given interval, we waited several seconds until the movement stopped before marking the video. We then raised the metal frame and poles to the water's surface, moved to the next subsample location, and repeated these steps for the remaining three video subsamples. While in the field, we recorded the 0.5-m interval at which each of the four video samples began. Based on this height, all subsequent heights could be determined by viewing the videos and noting the number of Highlight marks until the lake bottom was reached. Setup and sampling at each site typically took 15 to 25 min to complete (3 to 5 min per video sample), but this varied, primarily depending on weather conditions.

For winter sampling (January and February), we employed similar methods as described above but while standing on the ice. First, we located each sampling site and marked locations for two video samples located 5 m apart (the greater effort required for winter sampling made collection of four subsamples unfeasible). We then used an ice auger and ice saw to cut a hole through the ice large enough for the sampling device to pass through (approximately 1 by 0.75 m; Figure 2D) and recorded video samples in the same manner described above for the growing season. The mean depth at which we operated the sampling device was 1.6 m, with a maximum depth of 3 m; however, we have collected data with the device up to 3.9 m depth (W. Glisson, unpub. data) and it can be operated up to 5 m deep.

To determine whether data recorded from the videos (e.g., stem count) were a suitable proxy for biomass, we sampled biomass at the five lakes mentioned above and three additional lakes with watermilfoil beds (Lake Auburn, Carver County; Spectacle Lake, Isanti County; and Thomas Lake, Dakota County). We collected biomass at these lakes in October 2018 by randomly selecting one subsampling location at each site from which to collect biomass. After recording video data, we held the sampling device in place

and lowered a seven-tine (15-cm-wide) rake attached to a telescoping pole into the water such that it fell within the metal sampling frame (i.e., in front of the camera). We made two rotations with the rake and then pulled the rake and any attached biomass to the surface (vertical rake method following Johnson and Newman 2011). We repeated this procedure a second time within the metal sampling frame, adjacent to the first sample, to ensure that we collected all watermilfoil biomass that could be observed within the sampling window. We separated watermilfoil biomass from other vegetation, spun the biomass in a salad spinner for 30 to 60 s to remove excess water, and weighed the biomass in a plastic bag with a spring scale. This yielded a dataset of 40 subsample locations from which we had paired data for both wet biomass and video-based abundance measures.

Video and data analysis

We viewed the videos on a desktop computer with free and user-friendly GoPro Studio software. From each video, we determined the approximate height of watermilfoil plants (in 0.5-m increments) by recording the first 0.5-m interval at which a watermilfoil stem was observed within the 30 by 70-cm sampling window. Then, at each 0.5-m interval, we counted the number of clearly visible watermilfoil stems and estimated the vertical cover of watermilfoil in front of the sampling window (i.e., between the camera and the sampling window). We estimated vertical cover with arcsine-square root cover classes (> 0 to 1, > 1 to 5, > 5 to 25, > 25 to 50, > 50 to 75, > 75 to 95, > 95 to 99, > 99 to 100; Muir and McCune 1987).

Stem count and cover, like biomass, are measures of plant species' abundances, and these measures, along with plant height, could be characterized in several different ways. For example, each measure could be examined individually or combined into an aggregate metric (e.g., stem count \times stem height *sensu* Pine et al. 1989). Furthermore, data for each 0.5-m interval could be treated as a separate subsample (within each video sample) or summarized as cumulative, mean, or maximum values across intervals. In our data, watermilfoil stem count and cover at each 0.5-m interval were highly correlated ($r = 0.80$; $P < 0.001$). Thus, we chose to focus on stem count, which is simple to measure and easy to interpret biologically. Because we counted stems at each 0.5-m interval in our video samples, but collected a single corresponding biomass sample, we needed a way to combine multiple stem counts for each video sample into a single value for abundance. We also wanted to determine a measure of abundance that corresponded well with biomass; this necessitated a metric that incorporated both watermilfoil stems and the heights of those stems. Hence, we summed the stem counts taken at each 0.5-m interval from the water's surface to the substrate for each video sample. This resulted in a cumulative stem count metric, intended to reliably reflect abundance, which simultaneously incorporated both the number and heights of stems. For each of the four video samples at a site, we calculated this cumulative stem count metric. Across all sites and sampling events (June 2017 to February 2018), this generated 820 watermilfoil stem count values (Figure 4).

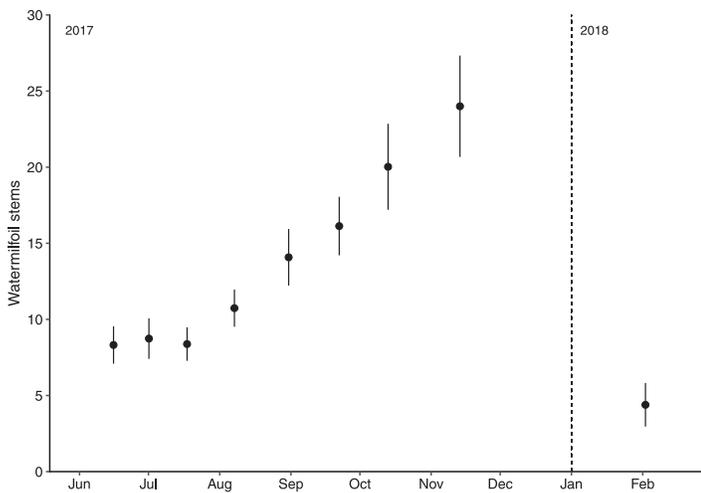


Figure 4. Watermilfoil stem count determined with the sampling device in five Minnesota lakes from June 2017 to February 2018. Points are mean cumulative stems \pm 1 SE from each video sample across all sites and lakes. Points are located at the mean date among all lakes for a given visit.

We examined the relationship between cumulative stem count and biomass with Pearson's correlation coefficient and a linear model in R version 3.4 (R Development Core Team 2017). For these analyses, we used the 40 paired cumulative stem count and wet biomass values. We log-transformed both biomass and cumulative stem count data to improve normality and homogeneity of variance. Because some samples had no watermilfoil, we added the minimum biomass value (1.5 g) to all biomass data and the minimum stem count value (1) to all cumulative stem count data prior to log transformation.

RESULTS AND DISCUSSION

Stems counted from video samples were strongly correlated with biomass recorded in the field ($r = 0.93$, $P < 0.001$; Figure 5), indicating that visual estimation using this sampling device was a good proxy for biomass. The seasonal growth pattern we observed with our nondestructive sampling device (Figure 4) was similar to patterns observed in other studies of watermilfoil in northern U.S. lakes that used destructive sampling (Adams and McCracken 1974, Perkins and Sytsma 1987). Hence, use of stem counts (or alternatively, cover) from video recordings appears to correspond well not only with snapshot measures of biomass, but also with seasonal changes in biomass. Moreover, we were able to observe plants and document watermilfoil abundance under the ice, which is generally prohibitive for diver-based sampling techniques (e.g., as in Adams and McCracken 1974, Perkins and Sytsma 1987). Importantly, the sampling device provided clear images of watermilfoil and other aquatic plants *in situ* (Figure 6). Overall, the sampling device appears to have good utility for measurement of biomass, growth, phenology, and morphology of watermilfoil without the need to destroy target and nontarget plants or enter the water. Transferability to other submersed macrophytes still needs to be evaluated, but this device is likely to at least be effective for other species with similar growth forms.

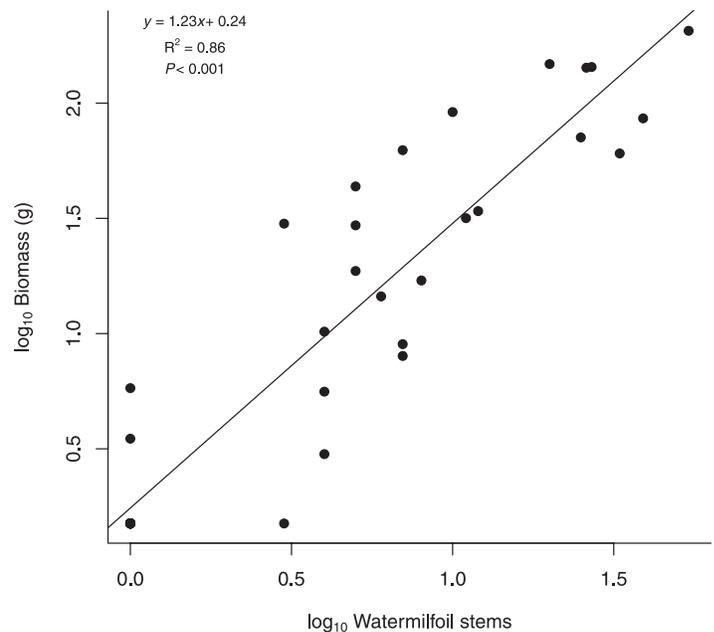


Figure 5. Relationship between watermilfoil stem counts measured from videos (cumulative stems per video sample) and wet biomass collected at the same sampling locations. The linear equation, R^2 , and P -value are from the linear model: $\log_{10}(\text{biomass} + 1.5) \sim \log_{10}(\text{watermilfoil stems} + 1.0)x + b$.

By providing clear images of aquatic plants and reasonable abundance estimates, this sampling device could be used for a variety of aquatic plant monitoring and research applications. This device is particularly well suited for seasonal or long-term monitoring of rare and threatened species' abundance and phenology. The ability to track aquatic plant growth and phenology throughout the year suggests that this approach could be extended to evaluate other changes in aquatic plant abundance over time, such as responses of invasive species to management. Effects of management on invasive species abundance could be determined, as well as the condition of treated plants, e.g., regrowth from herbicide-damaged or senesced stems, as has been observed by divers for Eurasian and hybrid watermilfoil (Thum et al. 2017). Indeed, we observed regrowth from senesced stems from the previous growing season in our videos. If permanent plots are sampled over time in such studies, use of our sampling device allows for repeated sampling without altering biomass of target plants or the plant community over time. Additionally, characterization of submersed plants' structure in the water column (e.g., height, growth form, and canopy position) observed with the sampling device can provide important information on the biology and ecology of aquatic plant communities.

Documenting the abundance, growth, and morphology of individual native and invasive species is likely the primary application for this device, but whole aquatic plant communities could also be examined. For example, point-intercept surveys typically conducted using thrown-rake abundance (Deppe and Lathrop 1992) or spun-rake biomass (Johnson and Newman 2011) methods could be implemented with our sampling device. Or the device could be used to augment rake-based methods to better account for species that are



Figure 6. Still images of watermilfoil captured from video taken with the sampling device. Estimated stem count and cover classes for the images: stems = 8, cover = 3 (top left); stems = 33, cover = 5 (top right); stems = 9, cover = 3 (bottom left); and stems = 1, cover = 2 (bottom right).

difficult to sample using rakes, such as low-growing or fragile species (Owens et al. 2010, Johnson and Newman 2011). The sampling device could also be used for scouting research locations (e.g., identifying areas where species of interest occur for establishment of experimental plots) or aquatic plant education and outreach (e.g., introducing lake groups or youth to the diversity of submersed aquatic plants). The use of the sampling device for any of these situations should be evaluated prior to implementation; modifications may be needed to ensure that project and sampling goals are met.

The design of the sampling device could be customized for different applications. For example, the camera could be positioned closer or farther away from the sampling window and the sampling window itself could be made smaller or larger, depending on the spatial scale of interest. The metal sampling frame or camera platform could also be constructed such that the camera faces down toward the lake bottom or up toward the surface to capture cover or canopy measures, respectively.

While the sampling device overcomes some limitations of traditional destructive and nondestructive sampling methods, it is not applicable to all situations. For example, translucent, inconspicuous, or diminutive species (e.g., *Utricularia gibba* L.) may be difficult to observe, especially under low water clarity. Despite the relatively close position of our camera to the

sampling window, observations were sometimes difficult for low-clarity lakes. Submersed plants that grow in dense beds or have a bushy growth form (e.g., *Chara* spp.) may also be difficult to assess as the sampling frame may press these plants down toward the substrate. Given the time it takes to deploy the sampling device, fewer samples could likely be collected in a given timeframe than traditional thrown- or spun-rake techniques. The device is nondestructive, but it is invasive and native plants were occasionally fragmented or uprooted by the frame and wire mesh. However, we expect that damage to plant communities will be minimal. Lastly, as with any field equipment, invasive species can attach to the sampling device. Care should be taken to ensure that any attached plants are removed and that the sampling device is thoroughly cleaned between sampling locations and lakes.

The underwater videos captured by our sampling device enabled us to measure watermilfoil abundance and examine growth and morphology in ways previously only attainable by destructive biomass sampling or nondestructive diver-based surveys. Recording underwater video with this sampling device from above the water's surface allows for year-round data collection, with minimal training investment for field staff. This sampling device is effective and easy to use, and can be tailored to the requirements of diverse research and monitoring applications.

SOURCES OF MATERIALS

¹Six-foot aluminum button bull float handle, Kraft Tool Co., 8325 Hedge Lane Terrace, Shawnee, KS 66227.

²GoPro HERO 5 Black, GoPro Inc., 3000 Clearview Way, San Mateo, CA 94402.

³iPad Air 2, Apple Inc., 1 Infinite Loop, Cupertino, CA 95014.

⁴Scotch Extremely Strong Fasteners, 3M, 2501 Hudson Road, Maplewood, MN 55144.

ACKNOWLEDGEMENTS

Funding was provided by the U.S. Army Corps of Engineers Research and Development Center. Michael Netherland provided vital support and guidance for the watermilfoil research that motivated development of this sampling device. Matthew Houser helped design the metal sampling frame and produced a technical drawing. William Hoffman built and welded the metal sampling frame. We thank Carli Wagner, Rafael Contreras-Rangel, Ranjan Muthukrishnan, Carolyn Kalinowski, and Noah Berg for field help.

LITERATURE CITED

- Adams MS, McCracken MD. 1974. Seasonal production of the *Myriophyllum* component of the littoral of Lake Wingra, Wisconsin. *J. Ecol.* 62:457–465.
- Brainard AS, Schulz KL. 2017. Impacts of the cryptic macroalgal invader, *Nitellopsis obtusa*, on macrophyte communities. *Freshw. Sci.* 36:55–62.
- Crowell W, Troelstrup N, Queen L, Perry J. 1994. Effects of harvesting on plant communities dominated by Eurasian watermilfoil in Lake Minnetonka, MN. *J. Aquat. Plant Manage.* 32:56–60.
- Deppe ER, Lathrop RC. 1992. A comparison of two rake sampling techniques for sampling aquatic macrophytes. Wisconsin Department of Natural Resources, Bureau of Research Findings (Number 32), Madison, WI.
- Glisson WJ, Wagner CK, McComas SR, Farnum K, Verhoeven MR, Muthukrishnan R, Larkin DJ. 2018. Response of the invasive alga starry stonewort (*Nitellopsis obtusa*) to control efforts in a Minnesota lake. *Lake Reserv. Manage.* 34:283–295.
- Johnson JA, Jones AR, Newman RM. 2012. Evaluation of lakewide, early season herbicide treatments for controlling invasive curlyleaf pondweed (*Potamogeton crispus*) in Minnesota lakes. *Lake Reserv. Manage.* 28:346–363.
- Johnson JA, Newman RM. 2011. A comparison of two methods for sampling biomass of aquatic plants. *J. Aquat. Plant Manage.* 49:1–8.
- Maceina MJ, Shireman JV, Langeland KA, Canfield DE. 1984. Prediction of submersed plant biomass by use of a recording fathometer. *J. Aquat. Plant Manage.* 22:35–38.
- Madsen JD. 1993. Biomass techniques for monitoring and assessing control of aquatic vegetation. *Lake Reserv. Manage.* 7:141–154.
- Madsen JD. 1997. Seasonal biomass and carbohydrate allocation in a southern population of Eurasian watermilfoil. *J. Aquat. Plant Manage.* 35:15–21.
- Madsen JD, Wersal RM. 2017. A review of aquatic plant monitoring and assessment methods. *J. Aquat. Plant Manage.* 55:1–12.
- Madsen JD, Wersal RM, Woolf TE. 2007. A new core sampler for estimating biomass of submersed aquatic macrophytes. *J. Aquat. Plant Manage.* 45:31–34.
- Marko MD, Madsen JD, Smith RA, Sartain B, Olson CL. 2015. Ecology and phenology of flowering rush in the Detroit Lakes Chain of Lakes, Minnesota. *J. Aquat. Plant Manage.* 53:54–63.
- Muir PS, McCune B. 1987. Index construction for foliar symptoms of air pollution injury. *Plant Dis.* 71:558–565.
- Owens CS, Smart RM, Williams PE, Spickard MR. 2010. Comparison of three biomass sampling techniques on submersed aquatic plants in a northern tier lake. U.S. Army Engineer Research and Development Center, APCRP Technical Notes Collection, Vicksburg, MS. 10 pp.
- Parks SR, McNair JN, Hausler P, Tynning P, Thum RA. 2016. Divergent responses of cryptic invasive watermilfoil to treatment with auxinic herbicides in a large Michigan lake. *Lake Reserv. Manage.* 32:366–372.
- Parsons JK, Couto A, Hamel KS, Marx GE. 2009. Effect of fluridone on macrophytes and fish in a coastal Washington lake. *J. Aquat. Plant Manage.* 47:31–40.
- Perkins MA, Sytsma MD. 1987. Harvesting and carbohydrate accumulation in Eurasian watermilfoil. *J. Aquat. Plant Manage.* 25:57–62.
- Pine RT, Anderson LWJ, Hung SSO. 1989. Non-destructive estimation of aquatic macrophyte biomass. *J. Aquat. Plant Manage.* 27:47–49.
- R Development Core Team. 2017. R: A language and environment for statistical computing. R Foundation for Statistical Computing, Vienna, Austria.
- Rothlisberger JD, Chadderton WL, McNulty J, Lodge DM. 2010. Aquatic invasive species transport via trailered boats: What is being moved, who is moving it, and what can be done. *Fisheries* 35:121–132.
- Sabol BM, Kannenberg J, Skogerboe JG. 2009. Integrating acoustic mapping into operational aquatic plant management: a case study in Wisconsin. *J. Aquat. Plant Manage.* 47:44–52.
- Sheldon RB, Boylen CW. 1978. An underwater survey method for estimating submersed macrophyte population density and biomass. *Aquat. Bot.* 4:65–72.
- Thum RA, Parks S, McNair JN, Tynning P, Hausler P, Chadderton L, Tucker A, Monfils A. 2017. Survival and vegetative regrowth of Eurasian and hybrid watermilfoil following operational treatment with auxinic herbicides in Gun Lake, Michigan. *J. Aquat. Plant Manage.* 55:103–107.
- Valley RD, Drake MT, Anderson CS. 2005. Evaluation of alternative interpolation techniques for the mapping of remotely-sensed submersed vegetation abundance. *Aquat. Bot.* 81:13–25.
- Valley RD, Johnson MB, Dustin DL, Jones KD, Lauenstein MR, Nawrocki J. 2015. Combining hydroacoustic and point-intercept survey methods to assess aquatic plant species abundance patterns and community dominance. *J. Aquat. Plant Manage.* 53:121–129.
- Wersal RM, Cheshier JC, Madsen JD, Gerard PD. 2011. Phenology, starch allocation, and environmental effects on *Myriophyllum aquaticum*. *Aquat. Bot.* 95:194–199.