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NOTES

Non-destructive Estimation of Aquatic Macrophyte Biomass

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INTRODUCTION

Biomass estimation is an important tool in aquatic plant research for studies such as species distribution and abundance, succession and assessment of weed management operations. Many reports describe methods used to estimate and sample biomass (Forsberg, 1959; Edwards and Moore, 1975; Smart and Barko, 1988; Cassani et al., 1988). However, all of these studies employed destructive sampling for measurement of biomass. Few studies have been done to develop non-destructive methods for estimating biomass. A recording fathometer has been used with fairly reliable results at depths greater than 2 meters in lakes in Florida (Maceina and Shireman, 1980). However, the use of this equipment can be expensive and would be difficult in relatively shallow water studies. Thus, it has become necessary to develop a fast, non-destructive method of biomass estimation.

MATERIALS AND METHODS

The present study compares three methods: (1) fresh weight, (2) volume, and (3) mean shoot length as means for non-destructive biomass estimates of aquatic plant biomass. Each of these methods was simple to use in a reasonable amount of time and did not require expensive, sophisticated equipment. Plants selected for analysis were sago pondweed (*Potomogeton pectinatus* L.), Eurasian watermilfoil (*Myriophyllum spicatum* L.), and American pondweed (*Potamogeton nodosus* Poir.).

Four canals were used, each 122 m long by 3 m wide, concrete lined, trapezoidal shaped in cross section and filled with well water. Two canals were flowing and two were static so that plant morphological changes induced by static and flowing conditions would be included. Flow velocity was controlled by a gate valve at the upstream end and was 0.67 m/s at the upstream end which decreased to 0.25 m/s at the downstream end.

Temperature, pH, conductivity, dissolved oxygen, total hardness, total alkalinity, and turbidity were measured at noon during each trial. A Ryan Model J, 180 day, 5-35 C recording thermograph (Peabody-Ryan Instruments, Redmond, WA) was placed on the bottom of the canals. A Hydrolab System 8000 (Hydrolab Corp., Austin, TX) was used to measure pH, conductivity, and dissolved oxygen. A Hach kit (Hach Co., Loveland, OR) was used to measure total hardness and total alkalinity. An HF Instruments model DRT 15 (HF Instruments, Ltd., Bolton, Ontario, Canada) was used for turbidity measurements.

Eight containers each of sago pondweed, Eurasian watermilfoil, and American pondweed were planted in 44 cm by 34 cm by 5 cm deep plastic dishpans, placed approximately 1.0 m apart in random species order in a canal section and allowed to acclimate to their respective experimental conditions (static or flowing water) for a month. Amount of soil (Yolo clayey loam) in each container was equalized as much as possible. Number of plant species that could be studied concurrently was limited due to the high number of replicated experimental units needed for statistical comparison.

For the first non-destructive biomass estimation method, total weight of container, wetted soil, and plants was measured with a Chatillion hanging balance with a capacity of 27.25 kg. The initial weight of container and wetted soil was subtracted from the weight of container, wetted soil, and plants leaving a value approximating plant fresh weight biomass. The volume method used volume displacement of container, soil, and plants submerged in a 400 liter glass aquarium graduated in 1 cm increments (1 cm = 2 liter displacement). Volume displacement of soil without plants was subtracted from that with plants to give a value that approximated the plants alone. The third method consisted of measuring shoot length of 25 randomly selected plants. The product of the mean length and the total number of shoots (x variable) was used to estimate fresh weight biomass (y variable) by linear regression equation.

At the start of each experiment, canals were partially drained and containers of each species were removed and biomass determined with each of the three methods. After measurement, plants within containers were cut off at the soil line, blotted dry, and weighed on a Mettler balance to determine actual fresh weight biomass. The experiment was conducted during November through January, 1986 and in May, 1987, thus giving 64 replicates for each method with each plant species. Data from both static and flowing canals were combined for each method and compared statistically using linear regression. Equations of the line as well as coefficients of determination (r^2), F- and P-values were calculated.

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TABLE 1. WATER QUALITY FROM NOVEMBER AND DECEMBER, AND MAY EXPERIMENTS IN STATIC VERSUS FLOWING CANAL CONDITIONS. SAMPLES WERE TAKEN FROM CANAL 3 (STATIC) AND CANAL 1 (FLOWING).

	Winter		Spring
	Nov 25	Dec 17	May 31
	Static canal		
Total alkalinity (mg/L)	230.0	232.0	220.0
Total hardness (mg/L)	216.0	214.0	198.0
Conductivity (mhos/cm)	500.0	496.0	479.0
Dissolved oxygen (ppm)	13.0	13.6	12.3
pH	7.0	7.0	9.0
Turbidity (NTU)	0.4	0.6	1.5
	Flowing canal		
Total alkalinity (mg/L)	226.0	234.0	222.0
Total hardness (mg/L)	208.0	222.0	204.0
Conductivity (mhos/cm)	481.0	490.0	498.0
Dissolved oxygen (ppm)	12.6	15.3	9.2
pH	7.4	7.1	8.8
Turbidity (NTU)	2.6	1.3	1.5

RESULTS AND DISCUSSION

Water quality variables (Table 1) were very similar for both static and flowing canals with the exception of turbidity. Variables changed from winter to spring similarly in both static and flowing canals. Differences in turbidity were greatest in winter with 0.44 versus 2.6 Nephelometric Turbidity Units (NTU) in static versus flowing canals, respectively. In the spring trial, turbidity was the same for both systems at 1.5 NTU.

Fresh weight biomass values that all non-destructive biomass estimation methods were regressed against (y variable) had the following means and standard deviations for Eurasian watermilfoil, sago pondweed, and American pondweed, respectively: 13.36 g ± 16.55 g, 14.69 g ± 25.94 g, and 34.92 g ± 13.31 g. The weight method had the following means and standard deviations for Eurasian watermilfoil, sago pondweed, and American pondweed, respectively: 95.39 g ± 399.60 g, 202.94 g ± 227.16 g, and 97.39 g ± 372.28 g. The weight method had coefficients of determination ranging from 0.002 to 0.24. F-values were less than 1.0 and P-values approached 1.0, giving no significance at the 95% level. This indicates no precision in using this method for estimating biomass of plants. The volume method had the following means and standard deviations for Eurasian watermilfoil and sago pondweed respectively: 0.91 l ± 1.28 l and 8.00 l ± 0.63 l. Similar to the weight method, the volume method had coefficients of determination that were very low (r^2 = 0.02 and 0.113) for sago pondweed and Eurasian watermilfoil, respectively. F-values were 0.6 and 4.6, respectively and P-values were 0.44 and 0.04, respectively showing no significance at the 95% level for sago pondweed but, however, giving good significance at the 95% level with Eurasian watermilfoil. American pondweed was excluded from statistical analysis due to problems with loss of floating leaf material when it was transferred into the aquarium. The mean shoot length method by plant count had the following means and standard deviations for Eurasian watermilfoil, sago pondweed, and American pondweed, respectively: 20.1 cm ± 3.82 cm by 148.07 plants ± 59.00 plants, 12.00 cm ± 3.75 cm by 19.58 plants ± 22.58 plants, and 12.88 cm ± 4.17 cm by 24.30 plants ± 6.87 plants. The mean shoot length method had very high coefficients of determination for all three plant species, ranging from 0.83 to 0.93. F-values ranged from 73.9 to 396.3 and P-values were 0.0001, giving significance higher than the 95% level. This level of correlation indicates a high precision for estimating biomass. Variations in r^2 and F-values may have been caused by plant morphological differences induced by static versus flowing canal conditions and winter versus spring water quality.

In weight and volume methods, soil loss due to excessive handling of the containers was the limiting factor in precision of measurement. When using the weight method, the wet soil would either spill when the container was lifted out of the canals or when containers were hung on the balance. When using the volume method, the soil loss was noticeable when the containers were lifted out of the canals and when they were put into the aquarium. If the soil in the containers could have been sealed to prevent spillage, perhaps more consistant results would have been obtained. With the mean shoot length method, the limiting factor was the accuracy of the total plant count. Algae clinging to the plants made shoot length measurements and plant counts difficult. Removal of most epiphytic, filimentous algae preceding the measurements solved this problem.

TABLE 2. COMPARISON OF THREE NON-DESTRUCTIVE METHODS USED TO ESTIMATE FRESH WEIGHT BIOMASS OF THREE AQUATIC PLANTS. EACH VARIABLE IS REGRESSED AGAINST FRESH WEIGHT (n=64 MEASUREMENTS/SPECIES).

Method	Plant species	y-intercept	slope (n)	Cofficient of Determination (r^2)	F	P
Weight	sago pondweed	14.23	0.003	0.002	0.891	0.3512
	watermilfoil	8.14	0.009	0.024	0.065	0.8004
	American pondweed	37.74	0.010	0.002	0.061	0.8076
Volume	sago pondweed	140.23	0.118	0.020	0.605	0.4426
	watermilfoil	10.25	–0.516	0.113	4.604	0.0387
	American pondweed	—[1]	—	—	—	—
Mean Shoot length by Plant count	sago pondweed	–4.21	0.037	0.830	87.815	0.0001
	watermilfoil	–1.21	0.058	0.860	73.897	0.0001
	American pondweed	–13.10	0.073	0.932	396.306	0.0001

[1]Measurements not taken during trial due to leaf loss during handling of plants.

In Maceina and Shireman (1980) where a fathometer was used to non-destructively estimate biomass, a calibration run was done where a small number of plants were clipped and weighed and this biomass value compared to recorded lake bottom tracing. Similarly, a calibration may be made by clipping of a small sample of plants, weighing and a comparison made with the product of the mean shoot length method. Lack of precision in biomass estimation due to seasonal plant morphological changes would thus be eliminated.

The results of this study suggest that measurements of mean shoot length and numbers of shoots can provide a good estimate of aboveground biomass without destructive harvesting.

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Nitrogen and Phosphorus Uptake and Release by the Blue-Green Alga *Microcoleus lyngbyaceus*

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INTRODUCTION

The blue-green filamentous alga *Microcoleus lyngbyaceus* (Kutzing) Crouan is an important component of the drift algae community in the Indian River lagoon, Florida (Benz, et al., 1979; Hall and Eiseman, 1981) and large scale natural blooms of this species have been reported (Humm, 1956; Phillips, 1963). A complete description of this alga's bloom sequence is presented by Blair and Meyer (1986). During bloom conditions, floating windrows of the alga, along with uprooted seagrasses produce rafts of considerable biomass. When these mats begin to decompose, local and dramatic increases in surface water ammonium and phosphate concentrations can occur (Zimmermann and Montgomery, 1984).

The purpose of this study was to characterize the ability of *M. lyngbyaceus* to remove nutrients from ambient and enriched waters, and also to follow the release of nutrients under dark conditions in an attempt to understand the impact of nutrient loading on this alga in the Indian River area.

MATERIALS AND METHODS

Samples of *M. lyngbyaceus* were collected in June 1985 from seagrass blades and the sediment surface in shallow (~ 1 m) water in the Indian River lagoon. After collection, the algae were maintained for 48 hours under ambient temperature and light conditions in a continuous flow-through water system containing unfiltered Indian River water (IRW). Aliquots of algae (2.5g wet weight) were placed in 350 ml glass jars containing 150 ml of unfiltered IRW; control jars (minus algae) contained only 150 ml of the unfiltered IRW. The algae and controls were then incubated at 25-26C under continuous fluorescent lighting (200, $\mu E \cdot m^{-2} \cdot s^{-1}$). Replicate jars containing the algae and one control were harvested at zero time and then at 2, 4, 8, 16 and 24 hours after zero time. A combined ammonium/phosphate enrichment series were also harvested in the same maner at the aforementioned times. To determine nutrient release rates, this same experiment using ambient and enriched water was performed simultaneously in the dark.

After harvesting the jars, a 50-ml water sample from each jar was filtered through a 25-mm GF/C filter and immediately frozen for later nutrient analyses. The remaining sample was filtered through a 47-mm GF/C filter and subsequently dried at 65C to determine the dry weight of the algae. Ammonium (NH_4), dissolved reactive phosphate (DRP) and nitrite + nitrate ($NO_2 + NO_3$) were determined on the filtrate using an AutoAnalyzer II system with modified Technicon procedures (Zimermann, et al., 1977).

Uptake and release rates of nutrients (R) were calculated using the formula:

$$R = \frac{(C(I) - C(X))\,(V)}{D\,(t)}$$

where: C(I) = initial concentration (μM)
C(X) = final concentration (μM)
t = time (h)
V = volume (1)
D = dry weight of algae in (g)
R = μmol g dry weight^{-1} h^{-1}

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