The significance of waterfowl feces as a source of nutrients to algae in a prairie wetland

Sara L. Purcell and L. Gordon Goldsborough
Department of Botany, University of Manitoba
Winnipeg, Manitoba R3T 2N2

Introduction

Recognition of the importance of waterfowl in aquatic ecosystems has been largely from the perspective of those wishing to foster waterfowl stocks. While this management-oriented view and its associated research has lead to new understanding of wetland ecosystems, such basic issues as the degree to which waterfowl contribute to the maintenance and development of wetland ecosystems as a whole remain unresolved. In particular, the role that waterfowl play in regulating plant communities in wetlands is uncertain. Clearly, large populations of grazing waterfowl can reduce plant biomass. Lauridsen *et al.* (1993), for example, noted that plant biomass in an area in a shallow European lake from which grazing coots (*Fulica atra*) were excluded was up to 6.5 times higher than in areas where coots were abundant. Similar observations were made by Wrubleski (1984) during his study of the impact of grazing ducks in an area of Delta Marsh, Manitoba. However, effects of waterfowl on vegetation are not wholly negative, as their feces and decomposing carcasses release nutrients which can stimulate plant growth (Gere and Andrikovics 1992). Moss and Leah (1982) studied an English wetland, described as being "guanotrophic", for which feces of black-headed gulls (*Larus ridibundus*) accounted for 53-72% of the total phosphorus (P) input. Snow geese (*Chen caerulescens caerulescens*) are known to make significant contributions to the nitrogen (N) budget of sub-arctic salt marshes that are their summer breeding ground (Bazely and Jefferies 1989). Recently, Manny *et al.* (1994) estimated that flocks of Canada geese (*Branta canadensis*) and mallard ducks (*Anas platyrhynchos*) contributed as much as 70% of the P budget of Wintergreen Lake, Michigan, USA. Decomposition of waterfowl carcasses is also an important contributor of nutrients; Parmenter and Lamarra (1991) found that dead pintail ducks (*Anas acutas*) lost 65% of their total N and 30% of their P over a period of 10 months.

Despite the fact that birds are abundant in Canadian wetlands, the relative importance of their feces to plant nutrition and growth in prairie wetlands has not been measured quantitatively. Observations made anecdotally during our other research projects at the Delta Marsh indicate that tern feces introduced accidentally into small enclosures can lead to marked increases in N and P levels (Goldsborough, unpublished data). Recent experiments in which analytical grade N and P were added to enclosures showed that nutrient enrichment can affect the quantity and composition of plants, leading to abundant surface mats of filamentous green algae (e.g., Campeau *et al.* 1994; Murkin *et al.* 1994; McDougal *et al.* in press). Concomitant effects on the invertebrate fauna were also noted (B. Hann, unpublished data). Therefore, it is reasonable to hypothesize that natural sources of N and P may be significant factors in regulating the Delta marsh ecosystem.

This study was designed to address the impact that waterfowl feces have on algal and plant production within *in situ* enclosures in Delta Marsh. Based on the results of our research in wetland enclosures enriched with inorganic N and P (McDougal *et al.* in press), results from preliminary laboratory microcosms to which feces were added, and a conceptual model for freshwater wetlands developed by Goldsborough and Robinson (1996), our hypotheses for this experiment were as follows:

- Mineralization of waterfowl feces releases abundant N and P, some of which is immediately bound up by wetland sediments and submersed macrophytes. However, a portion is made available to the water column for assimilation into algal biomass. This stimulation should be most evident at high, intermittent loading levels.
- Nutrients from waterfowl feces will alter the composition and relative abundance of the primary producer assemblage, leading to a shift in the wetland from an epiphyton-dominated "open" state to a metaphyton-dominated "sheltered" state. Consequent deterioration of invertebrate habitat under dense metaphyton cover will decrease invertebrate diversity, and decrease the abundance of phytophilous microcrustaceans and macro-invertebrates.

Methods

Enclosures

Ten enclosures, 5m x 5m in size, were constructed in Blind Channel in late May 1995; of these, six were used in the present experiment (Fig. 1). The enclosures were supported by high density foam blocks fastened under the 40 cm wide wooden walkways which allowed the enclosures to float just above the water surface. A translucent plastic curtain was suspended from each frame. At the base of each curtain was a metal rod embedded into the sediments in order to anchor the curtains to prevent lateral flow between the enclosures and the surrounding marsh. Water depth during the study was about 1 m so each enclosure enclosed approximately 20,000 L of marsh water. Gee-type minnow traps were placed in each enclosure once the curtains were deployed so the effects of potential secondary consumers were reduced.

Collection, mixing, and addition of feces

Fresh feces from Canada geese (*Branta canadensis*) and mallard ducks (*Anas platyrhynchos*) were collected from captive and wild flocks at the Delta Waterfowl and Wetlands Research Station between May and July 1995, and stored at -30°C until required for experimental additions. The feces was thawed and mixed to make a uniform moist slurry which was poured uniformly over the surface of the target enclosure. The slurry initially remained at the water surface but it was mixed into the water column using a wooden paddle. Four of the six experimental enclosures were pulsed (spiked) with the waterfowl feces slurry on 28 June 28 1995 and 21 July 1995. The experimental treatments were as follows: 1) high feces load comprising 19.23 g total P and 7.90 g total N per addition per enclosure, 2) low feces load comprising 1.92 g total P and 0.79 g total N per addition per enclosure, and 3) untreated controls. There were two replicate enclosures of each treatment. Feces loadings were chosen to approximate N and P loadings similar to those employed in earlier nutrient enrichment experiments; they were not intended to simulate natural waterfowl input levels.

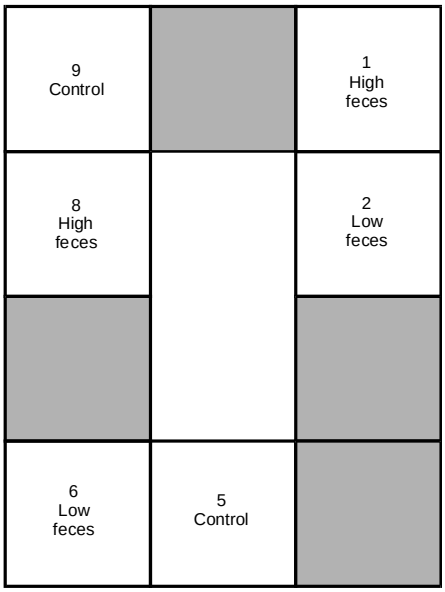

Figure 1. Schematic diagram of 5 x 5 m enclosures used in Blind Channel during 1995. Shaded enclosures were used in concurrent experiments (Higgins and Hann, this volume; McDougal and Goldsborough, this volume; Sandilands and Hann, this volume).

Sampling and Analysis

Measurements of light extinction, concentration of major nutrients, algal productivity (carbon assimilation), algal biomass (total chlorophyll), algal identification and macrophyte abundance were monitored in the six enclosures between June and August 1995. Light extinction was measured biweekly during bright, sunny days with a Li-Cor LI-185 meter and a submersible quantum sensor. Water samples were collected below the water surface and analyzed weekly for soluble reactive phosphorus (SRP), ammonia-N, soluble reactive silicon (Stainton *et al.* 1977), and nitrate-N (APHA 1992). Alkalinity and pH were measured concurrently with measurements of algal carbon assimilation (see below). Only data for SRP and ammonia-N are included in this report.

Phytoplankton were sampled at three randomly selected positions in each enclosure at weekly intervals. Four liters of enclosure water were collected using a plastic tube inserted vertically throughout the water column; the samples were filtered through a 100 µm mesh net to remove zooplankton. One liter of the filtrate was retained for analysis and the remainder was returned to the enclosure. Phytoplankton productivity (µgC/L/h) was determined from measuring the rate of carbon assimilation. Fifty milliliters were taken from each of the original 1 L water samples. It was dispensed in equal portions into two screw-cap test tubes, one of which was blackened with elastic tape. Each of the sub-samples were inoculated with a radiolabeled bicarbonate solution (0.5 µCi/mL) and placed in a 25°C water bath under saturating irradiance for four hours. The samples were then removed from the bath and collected onto glass fiber filters (Whatman GF/C) under gentle vacuum. The filters were placed over concentrated HCl for at least 1 minute, which volatilized any excess inorganic ^{14}C, and

placed into vials containing 5mL of Beckman ReadySafe™ liquid scintillation cocktail. The radioactivity of each vial was determined using a Beckman liquid scintillation counter. Phytoplankton biomass was determined from measuring its total chlorophyll content. Sub-samples (400mL) were taken from each of the original 1 L samples and collected onto glass fiber filters (Whatman GF/C) under vacuum. The filters were stored at -30°C for later analysis. Each filter was immersed in 5 mL of 90% methanol for 24 hours in the dark at room temperature. Each sample was then transferred into a cuvette and its absorbance was read at two wavelengths, 665nm and 750nm, with a Milton-Roy Spectronic 601 spectrophotometer against a blank of 90% methanol. Each sample was then injected with 50 µL of 10^{-3} M HCl, allowed to stand in the dark for one hour, then its absorbance was re-read at the same settings as before. The two sets of readings provided the information necessary to calculate the total chlorophyll content (µg/L) of each sample using the equation provided by Marker *et al.* (1980).

Additional phytoplankton sub-samples (200mL) were taken from each of the original 1 L samples, collected onto glass fiber filters (Whatman GF/C), and stored at 4°C for later analysis of total particulate phosphorus. The samples were combusted at 550°C in a muffle furnace for 1 hour, cooled, then injected with 25mL 1N HCl and boiled on a hot plate for 10 minutes. The samples were diluted to 100 mL with distilled deionized water and 10 mL were poured into glass vials where each sample received 2 mL of mixed molybdate reagent (4 parts acid molybdate to 1 part L-ascorbic acid). The samples were mixed and allowed to stand for 30 minutes to permit colour (blue) development. The samples were then transferred into glass cuvettes and the absorbance was read in a spectrophotometer at 885 nm against a blank of 10mL distilled water and 2mL mixed molybdate reagent. A standard curve for phosphorus was developed each time that samples were analyzed. The values obtained from the standard curve were graphed using a Lineweaver-Burke transformation from which we were able to calculate the concentration of phytoplankton particulate phosphorus (µgP/L). These data are not included in this report.

Sub-samples (100 mL) of the original 1 L sample were taken for subsequent algal identification and cell counts. Cells in the samples were allowed to settle overnight at 4°C then the majority of the water in the sample bottle was siphoned off, leaving the settled algal cells in approximately 10mL volume. This concentrated volume was centrifuged at 5,000 rpm for 10 minutes, after which 4 mL of the sample containing the algal cells were removed and poured into scintillation vials, each containing 1 mL of acid Lugol's solution. The samples were stored at 4°C for later identification and cell counts. Cells were counted at 400X using a 0.1 mL Palmer Cell on a Zeiss Axiolab phase contrast microscope. Phytoplankton from the following three divisions were identified using Prescott (1978) and counted: Cyanophyta, Chrysophyta (Bacillariophyceae only), and Chlorophyta. At 400X the field of view was 0.495 mm diameter and each field of view comprised one quadrat. Each sub-sample was counted, moving the stage from left to right and top to bottom until a total of 500 cells or 50 quadrats, whichever came first, was reached. Calculated algal density data (cells/L) are not included in this report.

Periphyton

Periphyton were sampled weekly using 90 cm long acrylic rods that served as artificial substrata for attached algae (Goldsborough *et al.* 1986). These rods were pre-notched at specific lengths, with each segment used for measurements of algal productivity, biomass, particulate phosphorus, or identification (Fig. 2). Sixty-four rods were positioned in early June so that an 8 x 8 grid was fashioned in each enclosure. The rods were not sampled for three weeks from the day they were placed in the enclosures. Three rods were sampled randomly from

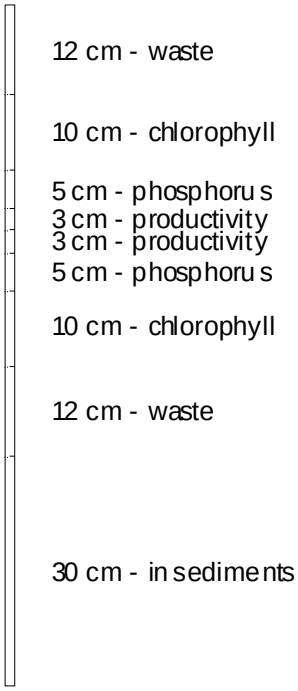

Figure 2. Schematic showing the segmentation of acrylic rods (0.64 cm diameter, 90 cm long) used as artificial substrata for periphytic algae in experimental enclosures in Delta Marsh.

each enclosure each week using a plastic tube that fastened around the rod while simultaneously taking a water column sample from around the rod. The water was removed carefully from the tube so as not to disturb the periphyton growing on the substratum. This water was filtered through a 100 µm plankton net for zooplankton analysis (Pettigrew and Hann 1996). The rods were snapped at the notched points with needle-nose pliers and the segments for total chlorophyll analysis (2 x 10 cm segments per rod), particulate organic phosphorus analysis (2 x 5 cm segments per rod), and algal identification (5 cm segment per rod) were placed in separate empty test tubes while the segments for productivity analysis (2 x 3 cm segments per rod) were placed in test tubes with 25 mL of pre-filtered marsh water previously taken from the enclosures.

Periphyton productivity (µg/cm^2/h), chlorophyll content (µg/cm^2), and particulate phosphorus content (µg/cm^2) were determined using the same basic procedures as performed for the phytoplankton samples. For productivity and chlorophyll measurements, intact rod segments were retained during analysis. For phosphorus analysis, periphytic algae were scraped from its substratum using a soft bristled paint brush into a petri dish containing a minimal amount of water. The contents of the petri dish were collected onto glass fiber filters (Whatman GF/C) for analysis. Phosphorus data are not reported here.

Rod segments preserved with acid Lugol's solution were stored at 4°C for later identification and cell counts. The vials were emptied into a petri plate and the rods were scraped with a rubber policeman. Sub-samples were taken from the contents on the petri plate and counted using the same procedure as for phytoplankton samples. Calculated algal censity data (cells/cm^2) are not included in this report.

Epipelon

Epipelon was sampled at three randomly selected sites in each enclosure at bi-weekly intervals. A small plastic tube, attached to a side-arm vacuum flask and a hand-operated vacuum pump, was used to suction up the surface sediments enclosed within a 10 cm diameter PVC tube that was embedded in the surface sediments in the enclosure. When the flask was filled, its contents were transferred into a 1 L sample bottle for transportation back to the laboratory. The contents of the plastic sample bottles were emptied into blackened 2L plastic bottles and left undisturbed in the dark for approximately 24 hours. Then, 400 mL of the overlying water of each sample were filtered through glass fiber filters (Whatman GF/C), from which 40 mL were taken for measurements of alkalinity and pH. Three flasks, each containing 100 mL of filtered water, were refrigerated overnight. The blackened beakers containing the sediment were transported to the UFS weather station. There, any overlying water was siphoned off and the sediment was covered with lens paper filters (Whatman #1), pre-cut to the same diameter as the beaker. The lens paper served to trap the upwardly migrating epipelon. Each beaker was covered with a clear plastic bag, held in place by an elastic band, and left undisturbed on the weather station platform overnight. The following morning (07:00), any excess water that had collected underneath the lens paper was carefully siphoned off with a glass pipette attached to the hand-operated vacuum pump. At 10:00, the lens papers were removed and placed into the corresponding 100 mL of filtered water. The flasks were shaken vigorously for 15 minutes to dislodge the epipelon trapped in the lens filter fibers. Then the water containing the suspended algal cells was used to measure epipelon carbon assimilation (µg/cm^2/h) and total chlorophyll (µg/cm^2) using the same procedures as used for phytoplankton samples.

Sediment

Two milliliters of fresh sediment were removed from each of the blackened beakers using a syringe to measure total sediment phosphorus content. The sediment samples were put into pre-weighed vials, dried at 100°C for 24 hours, and weighed. The samples were then processed using the same procedure as used for phytoplankton samples. Calculated phosphorus concentrations (µg/g) are not included in this report.

Macrophytes

Submersed macrophytes were sampled bi-weekly at three randomly selected positions in each enclosure, starting on 12 June. The sampler was modified from Pip and Stewart (1976) and enclosed all macrophytes contained in 0.09 m^2 of the enclosure bottom. Following collection, macrophytes were rinsed of phytophilous invertebrates using water from the enclosure, placed into a glass jar and shaken vigorously to dislodge epiphytes. The cleaned macrophyte samples were brought back to the lab where a sub-sample was taken to measure its surface area (length and diameter of stems, leaves). Small sub-samples, sorted by plant species, were placed in small pre-weighed aluminum dishes, dried at 100°C for 24 hours, and weighed. This allowed us to establish empirical relationships between the surface area (cm^2) and weight (g) of macrophyte species in the enclosures. Plant tissue not used in surface area measurements was

placed in pre-weighed aluminum plates and dried at 100°C for 24 hours, re-weighed, and used to calculate macrophyte biomass in the enclosure (g/m^2). A sub-sample of the dry macrophyte tissue was placed in pre-weighed glass vials for total phosphorus analysis (µg/g). These data are not reported here. The total volume of water used to clean the macrophytes, containing dislodged epiphyton, was recorded but only 1 L to was brought back to the lab for analysis of epiphyton biomass (total chlorophyll content). Known volumes (200 - 400 mL) of the sample were filtered (Whatman GF/C) for epiphyton chlorophyll ($µg/cm^2$) and total phosphorus ($µg/cm^2$) analyses. Phosphorus data are not included in this report.

Results

The nutrient concentrations in the water column for SRP (Fig. 3a) in the control and low organic load enclosures remained low throughout the sampling period. The high organic load enclosures, however, showed increased levels of SRP after each nutrient addition, particularly during August. Ammonia concentrations in the water column (Fig. 3b) peaked with each nutrient addition in the high organic load enclosures and continued to increase throughout August. The control and low organic load enclosures showed no difference in their ammonia concentrations.

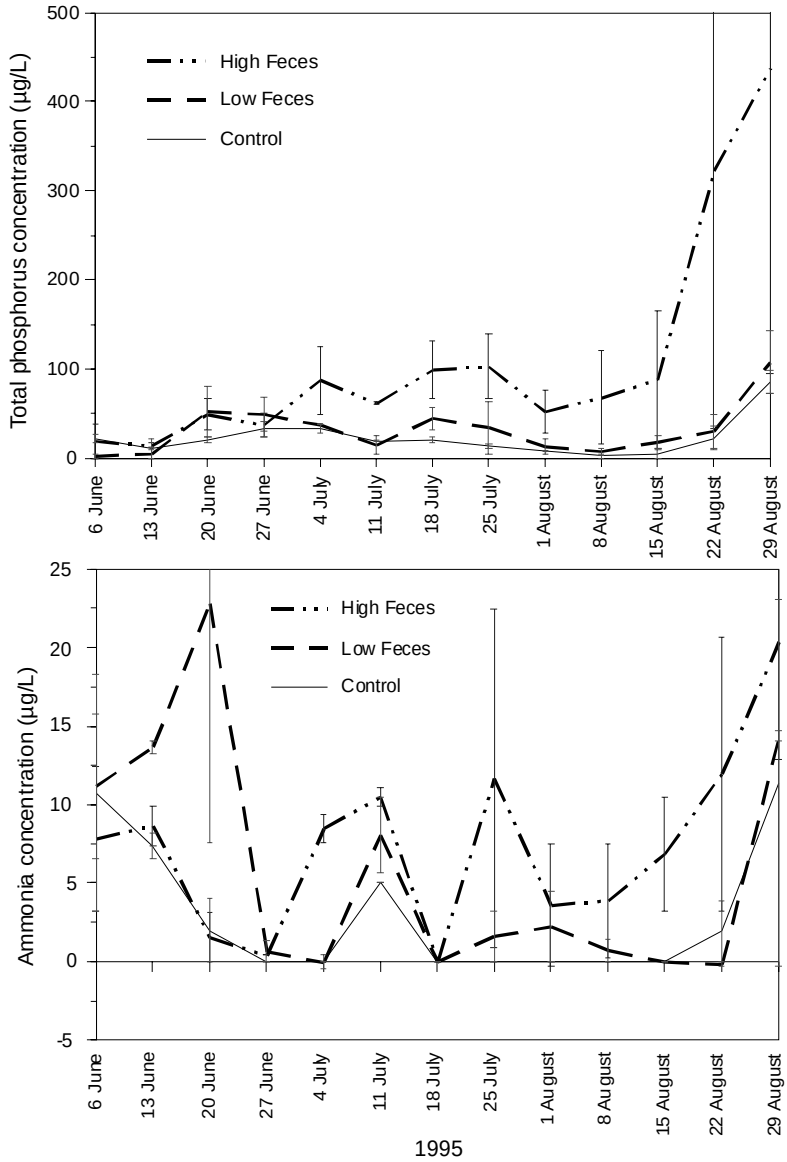

Figure 3. Changes in soluble reactive phosphorus (mg/L; top panel) and ammonia-N (mg/L; bottom panel) in the water column over a 13-week period in experimental enclosures. Additions of fresh waterfowl feces occurred on 28 June and 21 July.

Phytoplankton productivity (Fig. 4a) and chlorophyll concentrations (Fig. 4b) both increased after the first nutrient addition on 28 June in high organic load enclosures. However, this increase lasted only one week. Phytoplankton productivity remained low throughout the remainder of the sampling period in all three treatments. Phytoplankton chlorophyll concentrations increased through the month of August but there was no difference between the three treatments.

Periphyton productivity (Fig. 5a) and chlorophyll concentrations (Fig. 5b) show different trends. Periphyton productivity was essentially low in all three treatments with a slight difference between the high organic load enclosures and the other treatments towards the end of July. There are no differences observed in periphyton chlorophyll between the three treatments until after the second nutrient addition on 21 July. Then periphyton chlorophyll increased in the high and low organic load enclosures but not in the control enclosures. The increase in low organic load enclosures was short lived then remained low throughout the month of August. The control enclosures had consistently low levels of periphyton chlorophyll throughout the sampling period.

Epipelon productivity (Fig. 6a) and chlorophyll concentrations (Fig. 6b) showed no treatment effects. There were no differences between the three treatments throughout the sampling period.

Epiphyte chlorophyll (Fig. 7) increased steadily through the month of July in the high organic load enclosures. The controls and low organic load enclosures, however, had low epiphyte chlorophyll values throughout the sampling period with no difference between these treatments.

Metaphyton was absent from all enclosures throughout the experiment.

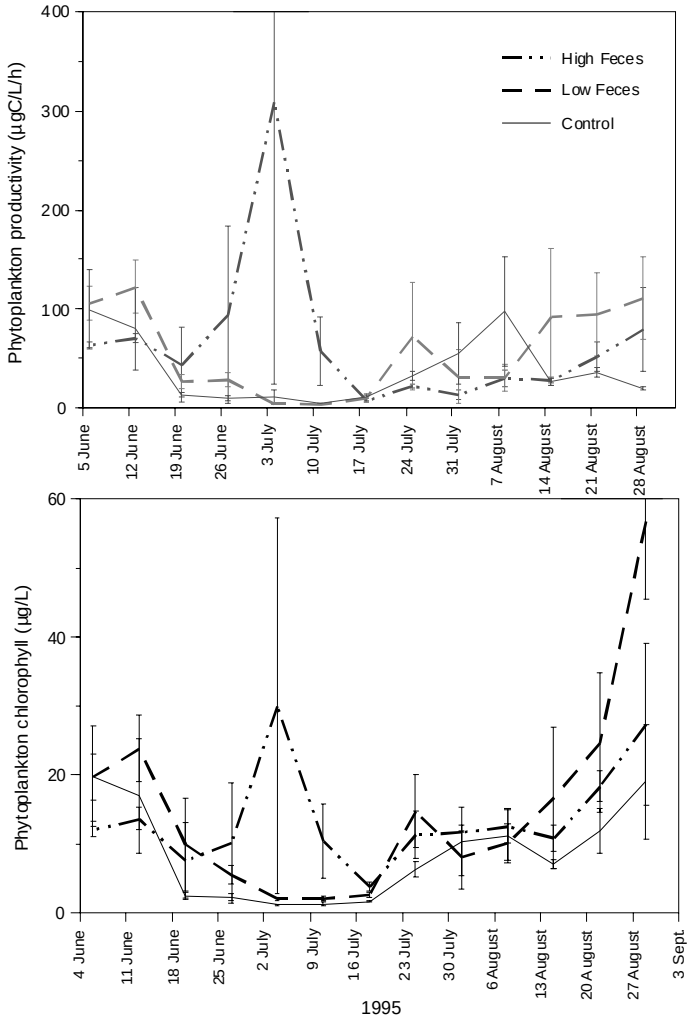

Figure 4. Changes in phytoplankton photosynthesis (top panel; µgC/L/h) and chlorophyll concentration (bottom panel; µg/L) over a 13-week period in experimental enclosures. Additions of fresh waterfowl feces occurred on 28 June and 21 July.

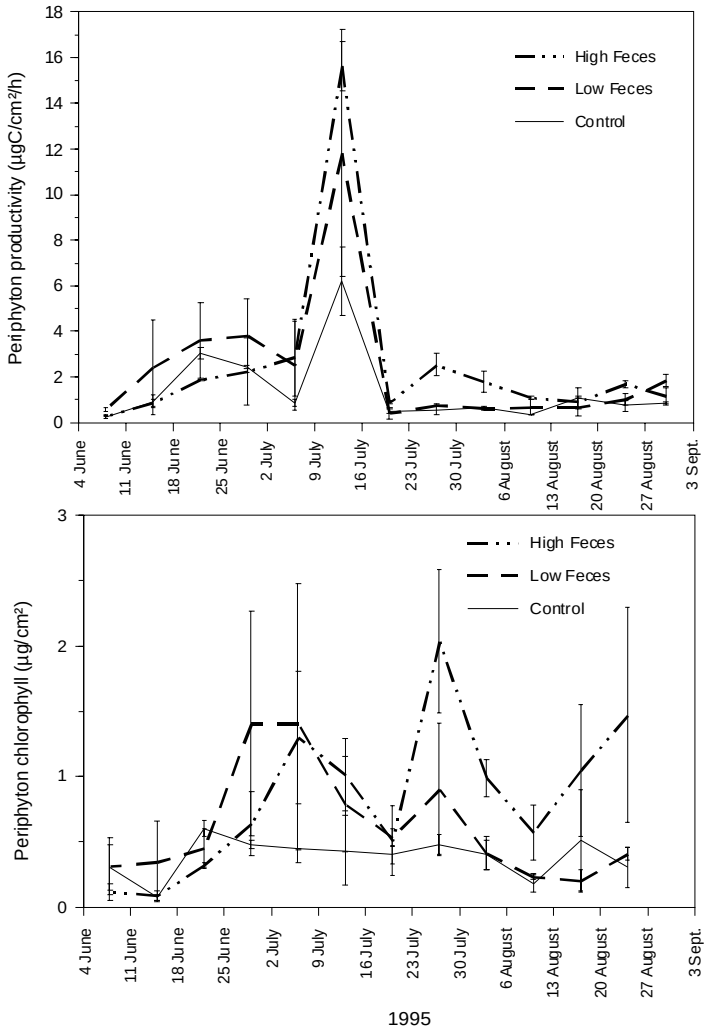

Figure 5. Changes in periphyton photosynthesis (top panel; µgC/cm²/h) and chlorophyll concentration (bottom panel; µg/cm²) over a 13-week period in experimental enclosures. Additions of fresh waterfowl feces occurred on 28 June and 21 July.

Discussion

Certain algal communities appear to respond to high concentrations of pulsed waterfowl feces additions during the growing season. The biomass (chlorophyll concentrations) response in high organic load enclosures as compared to the low organic load and controls appeared to be the most significant in the periphyton and epiphytes than responses by phytoplankton and epipelon. There was an increase in photosynthesis in the phytoplankton community after the first addition for approximately one week, however no response was observed when the second nutrient addition was added. Metaphyton mats, observed in inorganic nutrient additions (McDougal and Goldsborough 1995) were not observed with the addition of waterfowl feces.

Nutrient levels increased in the enriched enclosures as compared to the controls when the additions were applied. Nutrient levels continued to increase at the end of the sampling period. Comparison of the results from this experiment with those from laboratory aquaria enriched with waterfowl feces (Purcell, unpublished data) suggest this final increase in nutrient levels may be attributed to the absence (senescence) of macrophytes. The first addition of nutrients resulted in increased productivity and biomass of phytoplankton. At this period, the macrophytes had yet to become established. The second nutrient addition, when macrophytes was profuse, resulted in little change to phytoplankton productivity and biomass. This suggests that the second nutrient addition was utilized immediately by the macrophytes.

We did not observe a shift in community structure from an epiphyte-dominated system to a phytoplankton-dominated system. We propose that the reason for this was the abundance of macrophytes which were able to

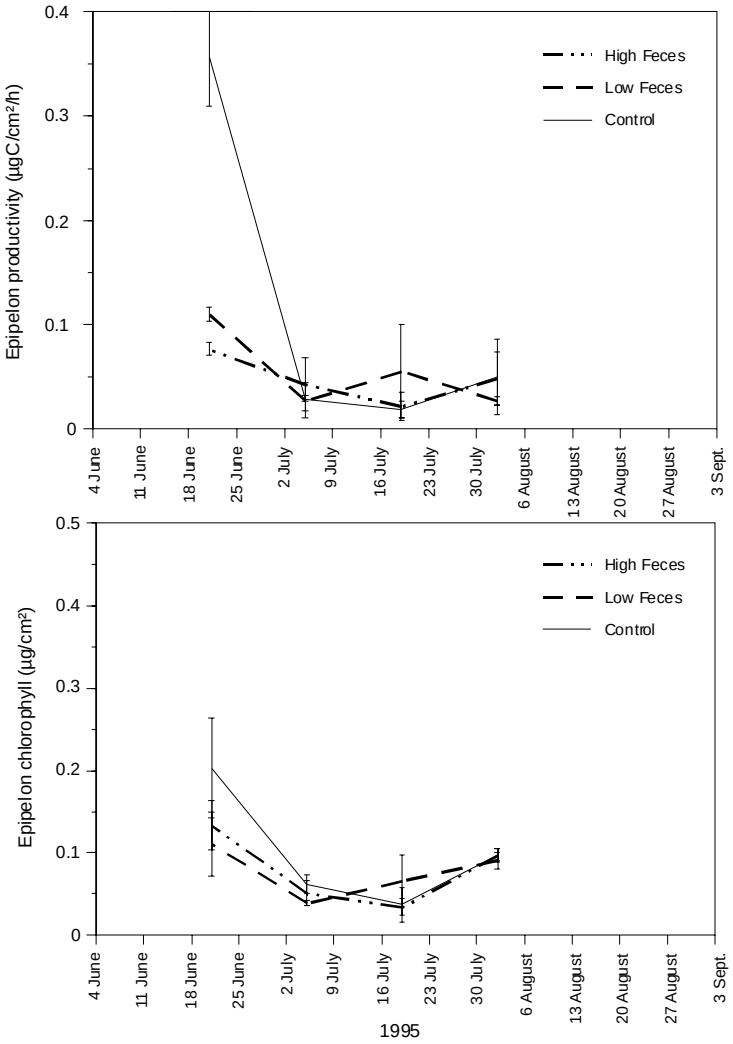

Figure 6. Changes in epipelon photosynthesis (top panel; µgC/cm^2/h) and chlorophyll concentration (bottom panel; µg/cm^2) over a 13-week period in experimental enclosures. Additions of fresh waterfowl feces occurred on 28 June and 21 July.

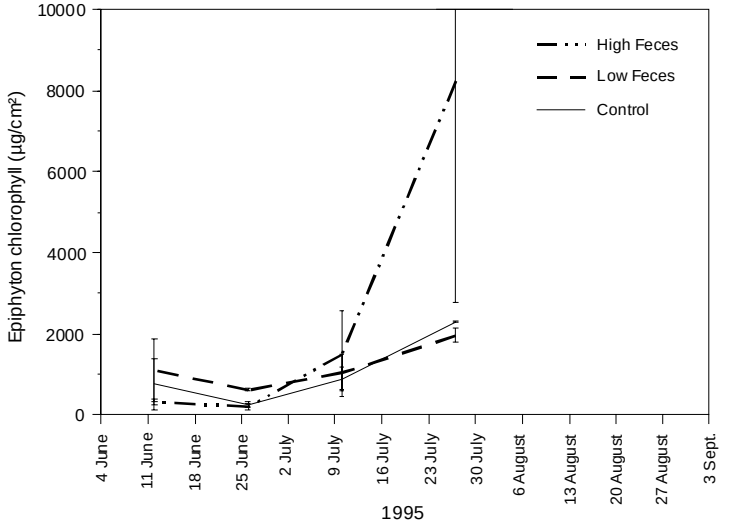

Figure 7. Changes in epiphyton chlorophyll concentration (µg/cm^2) over a 13-week period in experimental enclosures. Additions of fresh waterfowl feces occurred on 28 June and 21 July.

quickly assimilate nutrients released from the waterfowl feces, preventing their utilization by phytoplankton.

Acknowledgments

This study was funded by an NSERC Research Grant to Gordon Goldsborough. Assistance with sampling and analysis was provided by Rhonda McDougal. Field assistance was provided by Scott Higgins, Caedin Pettigrew, April Kiers, and Ken Sandilands. Special thanks to Curt Horning and Duane MacDonald for help collecting waterfowl feces.

References

American Public Health Association (APHA) 1992. Standard methods for the examination of water and wastewater, 18th edition. Greenberg, A.E., Clescerl, L.S. and Eaton, A.D. (eds.).

Bazely, D.R. and Jefferies, R.L. 1989. Lesser snow geese and the nitrogen economy of a grazed salt marsh. J. Ecol. **77**: 24-34.

Campeau, S., H. R. Murkin, and R. D. Titman. 1994. Relative importance of algae and emergent plant litter to freshwater marsh invertebrates. Can. J. Fish. Aq. Sci. **51**: 681-692.

Gere, G. and Andrikovics, S. 1992. Effects of waterfowl on water quality. Hydrobiologia **243/244**: 445-448.

Goldsborough, L.G. and Robinson, G.G.C. 1996. Pattern in wetlands. In: R. J. Stevenson, M. L. Bothwell and R. L. Lowe (eds.) Algal Ecology: Freshwater Benthic Ecosystems. Academic Press, pp. 77-117.

Goldsborough, L.G., Robinson, G.G.C. and Gurney, S.E. 1986. An enclosure/ substratum system for in situ ecological studies of periphyton. Arch. Hydrobiol. **106**: 373-393.

Lauridsen, T. L., E. Jeppesen, and F. Ø. Andersen. 1993. Colonization of submerged macrophytes in shallow fish manipulated Lake Væng: impact of sediment composition and waterfowl grazing. Aquatic Botany 46:1-15.

Manny, B.A., Johnson, W.C. and Wetzel, R.G. 1994. Nutrient additions by waterfowl to lakes and reservoirs - predicting their effects on productivity and water quality. Hydrobiologia **279/280**: 121-132.

Marker, A.F.H., Crowther, C.A. and Gunn, R.J.M. 1980. Methanol and acetone solvents for estimating chlorophyll a and phaeopigments by spectrophotometry. Arch. Hydrobiol. Beih. **14**: 52-69.

McDougal, R.L. and Goldsborough, L.G. 1996. The effect of macrophyte removal and inorganic nutrient addition on the algal communities in a prairie wetland. University Field Station (Delta Marsh) Annual Report **30**: 21-27.

Moss, B. and R. T. Leah. 1982. Changes in the ecosystem of a guanotrophic and brackish shallow lake in eastern England: potential problems in its restoration. *Internationale Revue gesamten Hydrobiologie* 67: 625-659.

Murkin, H. R., J. B. Pollard, M. P. Stainton, J. A. Boughen and R. D. Titman. 1994. Nutrient additions to wetlands in the Interlake region of Manitoba, Canada: effects of periodic additions throughout the growing season. *Hydrobiologia* 279/280:483-495.

Parmenter, R.R. and Lamarra, V.A. 1991. Nutrient cycling in freshwater marsh: the decomposition of fish and waterfowl carrion. Limnol. Oceanogr. **36**: 976-987

Pettigrew, C. and Hann, B.J. 1996. Cladoceran grazer response to pulsed organic nutrient additions in a freshwater marsh. University Field Station (Delta Marsh) Annual Report **30**: 52-62.

Pip, E. and Stewart, J. M. 1976. The dynamics of two aquatic plant-snail associations. Can. J. Zool. **54**: 1192-1205.

Prescott, G. W. 1978. How to know the freshwater algae. Wm. C. Brown Company Publishers, Dubuque, 293 pp.

Stainton, M.P., Capel, M.J. and Armstrong, F.A. 1977. The chemical analysis of freshwater. Second Edition. Fisheries and Environment Canada. Miscellaneous Special Publication No. 25.

Wrubleski, D. A. 1984. Species composition, emergence phenologies, and relative abundances of Chironomidae (Diptera) from the Delta Marsh, Manitoba, Canada. MSc thesis, University of Manitoba.