EUTROPHICATION AND NUTRIENT LOADING IN BARNEGAT BAY: IMPORTANCE OF SEDIMENT-WATER NUTRIENT INTERACTIONS

YEAR II

FINAL REPORT
Report No. 92-24F

Reference:
Research Agenda Item III. Coastal Research, A. Eutrophication, and C. Effects of Development Pressure on the Ecology of Barnegat Bay.

DSR Contact Person:
Mary Downes Gastrich, Ph.D.

Principal Investigator:
Sybil P. Seitzinger

Co-Investigator:
Isabel E. Pilling

Submitted:

22 October 1992

THE ACADEMY OF NATURAL SCIENCES

Division of Environmental Research
1900 Benjamin Franklin Parkway, Philadelphia, PA 19103

SUMMARY

Biological and chemical data collected in Barnegat Bay, NJ, during 1989 indicate that the Bay is in a moderately eutrophic condition. Phytoplankton production rates are considerably higher than rates in many other East Coast estuaries that receive substantial amounts of nutrient loading from pollutant sources. Phytoplankton production rates are greater in Barnegat Bay than in Narragansett Bay, Delaware Bay, Charleston Pond (a Rhode Island coastal lagoon), and Great South Bay, LI (a eutrophic coastal lagoon). Chlorophyll *a* concentrations are also high in Barnegat Bay compared to many estuaries including Upper Chesapeake Bay, Narragansett Bay and coastal lagoons in North Carolina, and are similar to chlorophyll concentrations in Great South Bay, LI. Light attenuation coefficients, and thus water column turbidities, are higher in Barnegat Bay than many other coastal lagoons. Detailed studies of factors controlling phytoplankton production in Barnegat Bay were not a part of the current study, but were investigated with nutrient enrichment studies using mesocosms during YR IV and will be presented in the YR IV Final Report.

The sediments in Barnegat Bay appear to be an efficient trap for nutrients (nitrogen, N and phosphorus, P) which are produced during decomposition of organic matter in the sediments. None of the phosphorus, and only a portion of the nitrogen, are recycled to the water column by diffusive flux. This contrasts with deeper estuaries where almost all of the phosphate and approximately half of the ammonia produced in the sediments, diffuses into the overlying water where it is taken up again by phytoplankton. The efficient nutrient trap in Barnegat Bay does not appear to be due to permanent burial in the sediments. Some of the nitrogen may be removed by denitrification in the sediments (not measured in current study).

Measurements of sediment-water nutrient fluxes and benthic primary production rates, as a function of light intensity in Barnegat Bay, suggest that benthic algae on the sediment surface are controlling the release of N from the sediments by assimilating ammonia as it diffuses across the sediment-water interface. In early summer and fall, when water column turbidity is lower and light levels at the sediment surface are relatively high, rates of benthic photosynthesis were substantial and no ammonia was released from the sediments. However, in mid-summer (July-August), when light levels were low at the sediment surface due to high water column turbidity, benthic photosynthesis was negligible and there was a substantial flux of ammonia out of the sediments. (Submerged aquatic vegetation, SAV, does not appear to be a major factor

controlling sediment-water nutrient fluxes at our study sites as patterns of N and P release were similar regardless of the presence or absence of SAV.)

While benthic algae are important in controlling sediment-water ammonia fluxes, the data indicate that factors in addition to benthic algae control phosphate fluxes. There was no measurable release of phosphate from the sediments regardless of whether benthic algal production rates were high or low. More extensive investigations of the mechanisms involved in sediment-water phosphate dynamics, including phosphate sorption to sediments, resuspension of bottom sediments such as may occur due to wave action or boating activity, and benthic algal interactions, were conducted during YR III and will be included in the YR III Final Report.

The removal of N and P in the sediments is currently decreasing the amount of N and P available for phytoplankton in the Bay and thus decreasing the apparent magnitude of eutrophication for the present rate of external nutrient inputs. However, an increase in external N and P inputs to the Bay would be expected to increase phytoplankton biomass, and thus, water column turbidity. If water column turbidities increase to the point that benthic algal production is no longer sufficient to assimilate N and P regenerated in the sediments, then the flux of N and P from the sediments to the water column would be expected to increase, leading to a further (and possibly non-linear) increase in phytoplankton biomass and production, and eutrophication in the Bay.

The efficient removal of P and N in the sediments contrasts markedly with data from deeper estuaries such as Narragansett Bay, Delaware Bay and Chesapeake Bay. In those estuaries, considerable amounts of N and P are recycled from the sediments to the water column. The findings of the current study suggest that models of nutrient control based on the relationship between nutrient inputs and eutrophication developed for deeper estuaries need to be modified for estuaries such as Barnegat Bay and likely for other shallow back bay estuaries or coastal lagoons.

TABLE OF CONTENTS

Page

SUMMARY i

LIST OF FIGURES iv

LIST OF TABLES vii

INTRODUCTION/BACKGROUND 1
General Discussion of Nutrient Cycling in Estuaries 2
Basic Theory of Sediment-Water Flux Measurements 3

METHODS 4
Water Column Nutrient Concentrations and Primary Production Rates 4
Sediment-Water Nutrient Fluxes 5
Study Areas 5
Nutrient and Oxygen Flux Measurements 5
Organic Matter Deposition Measurements 7
Sediment Trap Collections 7
N and P Burial in Sediments 9

RESULTS 10
Water Column Nutrient and Chlorophyll Concentrations and Primary Production Rates 10
Sediment-Water Nutrient and Oxygen Fluxes 11
Water Column Enclosures and Sediment Deposition Rates 12
N and P Burial in Sediments 13

DISCUSSION 15
Temporal and Spatial Variability of Water Quality 15
Factors Controlling Nutrient Availability and Eutrophication 16
Implications for Eutrophication in Barnegat Bay 18

LITERATURE CITED 20

FIGURES 24

TABLES 50

APPENDIX A 55

LIST OF FIGURES

Page

Figure 1. Map of Barnegat Bay, NJ 24

Figure 2. Water column enclosure 25

Figure 3. Nutrient concentrations at four locations in Barnegat Bay; A) ammonia, B) nitrate plus nitrite, and C) phosphate 26

Figure 4. Chlorophyll *a* concentrations 29

Figure 5. Secchi disk depths 30

Figure 6. Phytoplankton production rates (daytime gross prim. prod.) 31

Figure 7. Benthic metabolism in dark incubated cores--sandy vegetated site 33

Figure 8. Benthic metabolism in dark incubated cores--silt-clay site 34

Figure 9. Comparison of benthic metabolism in dark incubated cores --sandy vegetated and silt-clay sites 35

Figure 10. Comparison of benthic (gross) photosynthesis--sandy vegetated and silt-clay sites 36

Figure 11. Light profiles inside and outside the water column enclosures 37

Figure 12. Particulate P deposition at sandy vegetated site 38

Figure 13. Particulate N deposition at sandy vegetated site 39

Figure 14. Particulate C deposition at sandy vegetated site 40

LIST OF FIGURES (cont'd)

Page

Figure 15. Total and inorganic P concentrations at various depths --sandy vegetated site 41

Figure 16. Total Kjeldahl N and extractable ammonia concentrations at various depths--sandy vegetated site 42

Figure 17. Total and inorganic P concentrations at various depths --silt-clay site 43

Figure 18. Total Kjeldahl N and extractable ammonia concentrations at various depths--silt-clay site 44

Figure 19. Phytoplankton production rates in various estuaries during summer 45

Figure 20. Comparison of vertically averaged chlorophyll *a* concentrations in various estuaries 46

Figure 21. Light extinction coefficients in various estuaries during summer 47

Figure 22. Benthic photosynthesis (gross) versus ambient bottom light intensity--sandy vegetated and silt-clay sites 48

Figure 23. Sediment-water flux of ammonia vs. gross benthic photosynthesis 49

Appendix A

Figure A-1. A) Oxygen, B) phosphate, C) ammonia and D) nitrate + nitrite concentration versus time in water overlying laboratory-incubated sediment cores--sandy vegetated site, June 1989 55

LIST OF FIGURES (cont'd)

Page

Figure A-2. A) Oxygen, B) phosphate, C) ammonia and D) nitrate + nitrite concentration versus time in water overlying laboratory-incubated sediment cores--silt/clay site, June 1989 57

Figure A-3. A) Oxygen, B) phosphate, C) ammonia and D) nitrate + nitrite concentration versus time in water overlying laboratory-incubated sediment cores--sandy vegetated site, August 1989 59

Figure A-4. A) Oxygen, B) phosphate, C) ammonia and D) nitrate + nitrite concentration versus time in water overlying laboratory-incubated sediment cores--silt/clay site, August 1989 61

Figure A-5. A) Oxygen, B) phosphate, C) ammonia and D) nitrate + nitrite concentration versus time in water overlying laboratory-incubated sediment cores--sandy vegetated site, October 1989 63

Figure A-6. A) Oxygen, B) phosphate, C) ammonia and D) nitrate + nitrite concentration versus time in water overlying laboratory-incubated sediment cores--silt/clay site, October 1989 65

Figure A-7. A) Oxygen, B) phosphate, C) ammonia and D) nitrate + nitrite concentration versus time in water overlying sediments--*in situ* benthic chambers, sandy vegetated site, June 1989 67

LIST OF TABLES

Page

Table 1. Sediment-water fluxes of oxygen, ammonia, nitrite + nitrate and phosphate--sandy vegetated and silt-clay sites..... 50

Table 2. Number of phytoplankton in samples collected inside and outside enclosures 51

Table 3. Mid-day (gross) phytoplankton production inside and outside enclosures 52

Table 4. Comparison of conditions inside and outside water column enclosures..... 52

Table 5. Deposition of particulate nitrogen, phosphorus and carbon to sediments 53

Table 6. Sediment-water oxygen and nutrient fluxes in various estuaries 54

INTRODUCTION/BACKGROUND

Eutrophication[1] is a potential threat to the ecological health of Barnegat Bay and other shallow bays behind barrier islands (back bays or coastal lagoons) that line the New Jersey coast. Nutrients (nitrogen, N and phosphorus, P) enter these shallow bays from a variety of non-point and point sources (e.g., rivers and streams, storm drains, runoff, atmospheric deposition, broken sewer pipes, marinas and boating activity, groundwater, septic systems and direct discharge). Nutrient inputs to Barnegat Bay are expected to increase in the future due to continued population growth in the Bay watershed, as well as increased recreational use of the Bay by boaters.

Gauging the effects of increased development on eutrophication in Barnegat Bay is difficult, however, because there is little information on historic or current conditions. In addition, little is known about nutrient dynamics (including the relationship between nutrient inputs and eutrophication) in Barnegat Bay or any of these shallow, highly productive bays. Previous studies of eutrophication and nutrient dynamics in estuaries have focussed on relatively deep estuaries such as Delaware Bay, Narragansett Bay and Chesapeake Bay (D'Elia et al. 1986; Boynton et al. 1982; Nixon 1981). The extent to which results from studies of deeper estuaries can be used to predict the effects of nutrient inputs to shallow bays such as Barnegat Bay is unknown.

In order to assess current conditions in Barnegat Bay, as well as the possible effects on the Bay of increased development, the current study was conducted. The major objectives of the current study were to: 1) assess the temporal and spatial variability of water quality in Barnegat Bay, and 2) begin to quantify the important processes that control nutrient availability and thus eutrophication within the Bay, including the magnitude of the sediments as a source or removal site for nutrients.

To achieve these objectives the following measurements were conducted:

1) Water column inorganic nutrient concentrations (ammonia, nitrate plus nitrite, phosphate), chlorophyll *a* concentrations (New Jersey Department of Environmen-

1 Eutrophication results from high rates of nutrient (N and P) inputs to aquatic systems and can lead to a variety of conditions including algal blooms, increased water column turbidity, changes in species composition, and eventually to a depletion of oxygen in the water.

tal Protection and Energy; NJDEPE), turbidity (NJDEPE), and phytoplankton production rates (NJDEPE) were measured over an annual cycle at four stations throughout Barnegat Bay.

2) Sediment-water nutrient exchanges and oxygen fluxes were measured in early summer, late summer and fall at two sites in the highly developed northern portion of Barnegat Bay.

3) Deposition of new organic matter to the sediments was measured at the two benthic nutrient flux sites during summer and early fall.

4) Long-term burial of N and P in the sediments was measured at the two benthic nutrient flux sites.

A substantial focus on nutrient dynamics was directed at benthic processes because, as presented in detail in the Discussion, the sediments in many (deeper) estuaries have been shown to be an important source of recycled nutrients supporting phytoplankton production. In addition, the sediments can be an important site for nutrient removal. Understanding the magnitude of nutrient supply and/or removal in the sediments is, therefore, necessary to understanding factors that control algal production in the Bay.

General Discussion of Nutrient Cycling in Estuaries

Nutrients enter estuaries from a variety of external sources including river inputs, direct discharges, storm drains, runoff, atmospheric deposition, broken sewer pipes, marinas and boating activity, groundwater and septic systems. Once in the estuary the N and P are used by the algae for growth; however, the external inputs of nutrients to estuaries are generally not sufficient to supply the needs of algae. Recycling of nutrients within the water column and between the benthic sediments and water is a major source of N and P supporting algal production in estuaries (Boynton et al. 1982; Nixon 1981).

Inorganic nutrient (ammonia, nitrate and phosphate) inputs to the estuary are fixed into organic matter by phytoplankton, benthic algae and seagrasses. This organic matter is then consumed (metabolized) by microbes, zooplankton, fish, benthic invertebrates, etc., in the water column or sediments; eventually most is released back to the water column in the form of inorganic nutrients which can be used again by the algae and seagrasses. It is during the decomposition of organic matter in the sediments that N and P can be removed temporarily or permanently from the estuary, thus decreasing the supply of nutrients for phytoplankton production and affecting the degree of eutrophication resulting from a given rate of external nutrient input to the estuary.

Basic Theory of Sediment-Water Flux Measurements

In estuaries and other shallow marine systems, a considerable amount of the organic matter produced by phytoplankton or by benthic photosynthesis is decomposed (consumed, oxidized) in the bottom sediments by benthic organisms and bacteria. During the oxidation of organic matter in the presence of free oxygen, oxygen is consumed and carbon dioxide, ammonia or nitrate, and phosphate are released. According to Richards (1965), the decomposition of typical organic matter (carbohydrate) can be described stoichiometrically by the following equation:

$$(CH_2O)_{106}(NH_3)_{16}H_3PO_4 + 106O_2 = 106CO_2 + 16NH_3 + H_3PO_4 + 106H_2O \quad \text{(Eq. 1)}$$

According to this equation, 106 moles (212 atoms) of oxygen are consumed and 16 moles of nitrogen and 1 mole of phosphate are released for every mole of organic matter consumed. This ratio of C:O:N:P of 106:212:16:1 is referred to as the Redfield ratio for the decomposition of organic matter (Redfield 1934; 1958). The rate of oxygen consumption by benthic sediments can be used to estimate organic carbon decomposition, as well as the expected rate of release of ammonia and phosphate to the water column from the sediments due to organic matter decomposition (Nixon 1981; Boynton and Kemp 1985; Hopkinson and Wetzel 1982).

The equation for photosynthesis is essentially the reverse of the one for decomposition:

$$106CO_2 + 16NH_3 + H_3PO_4 + 106H_2O = (CH_2O)_{106}(NH_3)_{16}H_3PO_4 + 106O_2 \quad \text{(Eq. 2)}$$

Thus, for every 106 moles of carbon dioxide that phytoplankton or benthic algae and seagrasses fix into organic matter during photosynthesis, 16 moles of ammonia and 1 mole of phosphate are consumed and 106 moles (212 atoms) of oxygen are produced. This equation can be used to estimate the amount of ammonia and phosphate required to support measured rates of photosynthesis (rates of oxygen production). The importance of benthic processes in supplying phytoplankton nutrient requirements can be examined from a comparison of measured benthic N and P effluxes and calculated phytoplankton N and P requirements.

METHODS

Water Column Nutrient Concentrations and Primary Production Rates

Water column concentrations of ammonia, nitrate plus nitrite, and phosphate were measured at four locations extending from the less developed southern to the highly developed northern end of the Bay (Fig. 1) from April 1989 to April 1990. Nutrient samples were composite depth samples collected at surface, mid and near bottom depths by the NJDEPE DSR. Samples were stored on ice in the dark for transport to the Academy of Natural Sciences of Philadelphia (ANSP) where they were processed and then analyzed by ANSP. Ammonia samples were analyzed immediately (Solorzano 1969). Nitrate plus nitrite and phosphate samples were filtered with acid-cleaned glassware through pre-rinsed glass microfiber filters (Whatman 934-AH) and frozen for later analysis (modified from Technicon 1977; Murphy and Riley 1962). Primary production rates, chlorophyll *a* concentrations, and secchi disk depths were measured by the NJDEPE at the same locations, but at less frequent intervals. Primary production rates were calculated from O_2 changes in 250-ml BOD bottles incubated in the light or in the dark for 4 h at approximately 150 μEinsteins m^{-2} s^{-1} in a light box. O_2 concentrations were determined by Winkler titration (Carpenter 1965). Daily phytoplankton production rates (DPP) were calculated according to Keefe et al. (1981) as:

$$\text{DPP mmol C } m^{-2}d^{-1} = \text{mmol } O_2\ m^{-3}h^{-1} \times (0.5 \times D_1) \times 0.8 \times Hrs_{RS} \qquad \text{(Eq. 3)}$$

where D_1 is the depth of the 1% light level and Hrs_{RS} is the number of hours between sunrise and sunset. D_1 was calculated as $I_0 = I_z e^{-Kz}$, where I_0 and I_z are the light intensity at the surface and at the depth of the 1% light level, respectively, and K is the light extinction coefficient calculated from the measured secchi disk depth (D_s) as $K = 1.7/D_s$ (Parsons et al. 1977).

Sediment-Water Nutrient Fluxes

Study Areas

Sediments along the eastern (barrier island) portion of the Bay are primarily sandy, with finer grained silt-clay sediments in the deeper portions of the Bay and along the mainland side. Sediment-water nutrient and oxygen fluxes were measured in June, August and October 1989 at two locations in the highly developed northern end of Barnegat Bay: one fine grained silt-clay sediment area (~2 m water depth) and one sandy sediment area (with benthic algae and at times *Zostera* and/or *Ruppia*) (~1.2 m water depth) (Fig. 1). Both of these sites were included in the previous year's study (Seitzinger and Pilling 1989). Field-collected cores were incubated in the laboratory under ambient field light and temperatures in an environmental chamber. In addition, *in situ* and laboratory methods were compared as discussed below.

Nutrient and Oxygen Flux Measurements

Replicate sediment cores (17 cm diameter; approx. 15 cm deep) were collected by SCUBA-equipped divers using plastic coring tubes for benthic nutrient and oxygen flux measurements. Care was taken during coring to avoid disturbance of the sediment surface. At the time of sediment collection, water temperature (thermometer) and light intensity (LiCorr Model 185) at the sediment surface were measured, and water was collected from each location in carboys. The cores were maintained with overlying water at ambient bay water temperature during transport to the laboratory. The oxygen in the water over the cores was maintained at near saturated concentrations with the use of portable aerators. Once in the laboratory, the sediment cores were maintained at ambient bay water temperature in a temperature and light controlled environmental room. The water over all cores was continuously aerated and mixed by a gentle stream of air up until the time of the flux measurements.

The sediment-water exchanges of ammonia, nitrite plus nitrate, phosphate and oxygen were measured on replicate cores from each site (six per site in June and August; four per site in October) as follows (Seitzinger 1987a,b). Approximately 48 h after the sediment cores were collected, the water (approximately 2.5 L) over each core was changed with water collected from the site of sediment collection. The cores were aerated until approximately 1 h before the first nutrient samples were collected or oxygen measurements made. Approximately 1, 3, 6 and occasionally 24 h after the water was changed, samples of the overlying water were collected for nutrient analysis, after manually stirring the water with a small paddle. Oxygen concentrations in the overlying water were also measured using a YSI oxygen probe. The probe and meter were calibrated prior to the first series of samples and the calibration was verified with each sample interval. Oxygen levels were monitored frequently, and when necessary, the water was reaerated to prevent oxygen levels in the overlying water from dropping below 4 mg O_2 L^{-1}.

Two cores were maintained at ambient light levels (at the sediment surface at mid-day in the field), two at 50% ambient mid-day light levels, and two in the dark, for the

June and August experiments. During the October measurements, two cores were incubated at ambient field light conditions and two in the dark. In addition to chamber incubations, a series of light, medium and dark beakers was filled with bay water from the site of sediment collection and used as controls for assessing changes in nutrient concentrations in the water over the sediments due to water column processes. These controls were sampled at the same time intervals, using the same methods, as those specified above for the chambers.

Ammonia samples were analyzed immediately after collection (Solorzano 1969). Nitrate plus nitrite and phosphate samples were filtered immediately after collection with acid-cleaned glassware and prerinsed glass microfiber filters (Whatman 934-AH) and frozen for later analysis (modified from Technicon 1977; Murphy and Riley 1962).

Sediment-water nutrient and oxygen fluxes were calculated based on the time rate of change in the concentration of nutrients or oxygen in the water overlying the cores after correcting for concentration changes in control bottles, the volume of water over the cores, and the surface area of sediment as follows:

$$\text{Flux } (\mu\text{mole } m^{-2}h^{-1}) = \frac{(C - c) \times V}{A} \qquad \text{(Eq. 4)}$$

where C and c are the time rates of change in concentration in the water over the cores (C) or in control (c) bottles calculated from linear regression analysis of the data in units of μmol/L/h, V equals volume of water over cores in liters, A equals cross-sectional area of chamber in m^2.

Sediment-water oxygen fluxes in cores incubated in the light and dark were used to calculate rates of benthic organic matter decomposition and benthic photosynthesis as follows:

Organic matter decomposition rate = dark O_2 consumption rate (Eq. 5)

Net benthic photosynthesis = light O_2 production (Eq. 6)

Gross (total) benthic photosynthesis = light O_2 production + dark O_2 consumption (Eq. 7)

The calculated net benthic primary production is the net production of new organic matter in the sediments (net due to algal production and algal and animal respiration). The gross or total benthic photosynthesis is the sum of the net benthic photosynthesis plus the amount of oxygen that is being consumed in the light by algal and animal respiration (dark O_2 consumption).

In June, laboratory measurements of benthic nutrient and oxygen fluxes were compared with *in situ* measurements at the sandy vegetated site. *In situ* measurements were made the same day the cores were collected for the laboratory measurements. For the *in situ* incubations six chambers (12.5 cm diameter), including three light and three dark chambers, were placed over the bottom sediments for approximately 6 h.

Chambers were carefully deployed by divers using SCUBA gear so that bottom sediments remained as undisturbed as possible. Time series samples for nutrients were taken at approximately 1, 3 and 6 h. Oxygen levels were monitored using a YSI field oxygen meter and probe. An expansion glove inside the chamber allowed water samples for nutrient analysis to be withdrawn from the sealed chambers without admitting surrounding bay water or sediment pore waters (Seitzinger and Pilling 1990). The water inside the chambers was carefully and thoroughly stirred manually with a small paddle stirrer located inside each chamber before each sample was taken. In addition to chamber incubations, three light and three dark bottles were filled with bay water and were incubated on the bottom near the chambers and served as controls for assessing changes in nutrient concentrations due to water column processes.

Samples were kept in the dark on ice during transport to the laboratory and were analyzed according to procedures described above for laboratory benthic flux measurements.

Organic Matter Deposition Measurements

Sediment Trap Collections

The quantity of organic N and P deposited in the sediments was measured using sediment traps. Sediment traps have been used extensively to measure the vertical flux of particulate matter. A number of studies have evaluated the efficiency of various designs (Hargrave and Burns 1979; Bloesch and Burns 1980; Gardner 1980). The major problem with the use of sediment traps in estuaries is that they collect not only newly deposited material but also resuspended material. Resuspension often greatly exceeds the newly deposited material. (Hargrave and Taguchi 1978; Oviatt and Nixon 1975).

In order to overcome the resuspension problem, we isolated columns of water from resuspension for short periods of time and deployed sediment traps inside these enclosures. The objective of the enclosures is to isolate a column of water from further resuspension of bottom sediments long enough to measure new deposition of organic P and N, and briefly enough to avoid measurably altering the natural rate of organic sedimentation. The assumption is that the rate of sedimentation of newly deposited (not resuspended) organic matter inside the enclosures is the same as that outside the enclosures, if the primary production rate and phytoplankton species and abundances are similar in both. These sediment traps and enclosures underwent preliminary testing in Barnegat Bay during 1988; further testing was conducted in 1989.

The water column enclosures are cylindrical (1.0 m diameter), closed at the bottom, and made of clear, 10- to 12-ml reinforced polyethylene plastic sheeting with hoop supports at 0.5-m intervals (Fig. 2). A collar extends 15 cm above the surface of the water to prevent outside water from entering. The enclosures were filled by vertically collapsing the enclosure at the surface and then slowly lowering the anchor which was attached to the enclosure bottom to the sediment surface. The floatation ring was

simultaneously depressed below the surface of the water to allow the water to enter over the top. Cylindrical sediment traps with a height-to-diameter ratio of 5 were used to minimize resuspension in the traps (Bloesch and Burns 1980; Hargrave and Burns 1979). The traps were located away from the walls of the enclosure and as close to the bottom of the enclosure as possible to capture the organic matter which would have reached the sediment.

Last year the enclosures were open but anchored on the bottom (Seitzinger and Pilling 1990). There was concern that there might be some exchange of water either through the sediments or the bottom of the enclosure. To test this, we performed dye experiments using rose bengal. Time series samples were taken after the dye was added to the bag. Dye concentrations decreased with time indicating that either the bag leaked or that the dye was sorbing onto organic particles within the water column which were falling out of suspension. We therefore performed further tests with NaCl to differentiate between sorption and leakage. Tests carried out on two separate occasions demonstrated that there was mixing of water inside and outside the bag, presumably through the sediments. As a result of those tests the bag was closed off at the bottom with a drawstring and anchored to the bay bottom. Salinity experiments were again carried out and demonstrated that the salinity remained constant. For the 1989 field season, the water column enclosures were closed off at the bottom with a drawstring to prevent possible contamination of sediment trap samples.

The frequency and location of water column enclosure deployments were: 10 July and 1 August, two enclosures at the vegetated site; 8 and 11 August, one enclosure at the sandy site and one at the silt-clay site; 28 August, one enclosure at the silt-clay site; 5 September, two enclosures at each site; 14 September, four enclosures at the sandy site; 18 and 25 September, two enclosures at each site.

At least 48 h after enclosure deployment, sediment traps were deployed inside and outside the enclosures. Approximately 24 h after trap deployment the sediment trap samples were retrieved from inside and outside the enclosures. The entire contents of each sediment trap sample were decanted into a plastic bottle and stored in the dark on ice. Sediment trap samples were processed by filtering a known volume of sample through prerinsed and precombusted Whatman glass fibre filters (934-AH). The filters were dried (60°C) and frozen for later analysis of carbon (C) and nitrogen (N) using a Carlo Erba Model 1106 CHN analyzer. Replicate filters were analyzed for total particulate phosphorus (P) by persulfate digestion (Martin and Knauer 1973) followed by colorimetric analysis (Murphy and Riley 1962). N and P deposition rates were calculated based on the total N or P in the sediment trap (μmol), the surface area of the trap (A), and the trap deployment interval (HR) as:

$$\text{Deposition } (\mu\text{mol m}^{-2}\text{h}^{-1}) = \mu\text{mol}/(\text{A}^*\text{HR}) \qquad \text{(Eq. 8)}$$

Primary production rates, phytoplankton species and abundances, and nutrient concentrations inside and outside the two enclosures were compared during the 10 July deployment. Phytoplankton samples (whole water samples, 500 ml) were collected 48 h after enclosure deployment ~0.5 m below the surface of the water

inside each enclosure and outside the enclosures. Samples were preserved and stained with acid Lugols solution and stored in the dark on ice. Phytoplankton samples were processed by sedimentation and centrifugation (final concentration was approximately 50x). Representative samples of phytoplankton cells were counted and identified using a Palmer-Maloney nannoplankton counting cell with a compound microscope (approximately 400x magnification). Water samples were also collected inside and outside the enclosures for nutrient analysis. Nutrient samples were analyzed as described above for benthic flux studies.

N and P Burial in Sediments

Sediment cores (7 cm diameter; approx. 1 m deep) were collected from the sandy and silt-clay sites on 18 July 1990 by SCUBA-equipped divers. Cores were sectioned at 0.5-cm intervals for the first 5 cm, 1-cm intervals for the next 5 cm, and at 2-cm intervals for the remainder of the core. Inorganic phosphorous content of the sediments was determined based on the dissolved phosphate concentrations in subsamples (0.3 g dry wt.) extracted overnight with 1 N HCL (Froelich et al. 1988). Total P content was determined on sediment subsamples combusted at 450°C overnight to release organically bound phosphorus and then extracted with 1 N HCL (Froelich et al. 1988). Organically bound phosphorus was calculated as the difference between total and inorganic P. Total N content of the sediments was determined following semi-micro Kjeldahl digestion (APHA, AWWA and WPCF 1989). Sediment density was calculated for each depth interval based on wet and dry weight measurements of a known volume of sediment. Vertical profiles of ^{137}Cs and ^{7}Be were measured by Dr. Richard Bopp on replicate cores collected with a hand-held corer operated from the boat at the silt-clay site and used to calculate net sediment accumulation rates at that site.

Net burial of N and P in the sediments was calculated based on the estimated net sediment accumulation rate (cm/y), the measured sediment density, and the N and P content of the sediment (mg N or P/g dry sediment).

RESULTS

Water Column Nutrient and Chlorophyll Concentrations and Primary Production Rates

Water column concentrations of ammonia, nitrate plus nitrite, and phosphate were generally low at all four stations in the Bay from April through November 1989 (Figs. 3a-c). During the winter months samples were only collected at Mantoloking and Manahawkin. Ammonia concentrations were generally less than 2.5 μM and showed no strong seasonal pattern (Fig. 3a). Higher concentrations were noted on two occasions at the northern-most site (Mantoloking) and once at a mid-Bay station (Waretown). Nitrate plus nitrite concentrations (hereafter referred to as nitrate) were generally less than 2 μM (Fig. 3b). However, more variable and higher concentrations were noted frequently at Mantoloking in fall through winter and occasionally at other locations. Phosphate concentrations were less than 0.50 μM at all locations. There was a weak seasonal pattern at all stations of lower concentrations in spring through mid-summer, increased concentrations in mid- to late summer and then decreased concentrations again in late fall (Fig. 3c).

Chlorophyll concentrations were lowest in April, increased steadily to 20-28 mg/m^3 by July, and then decreased throughout the late summer until November (Fig. 4) (NJDEPE Leed's Point Laboratory, unpubl. data). Concentrations were generally lowest at Waretown, the station closest to the ocean inlet.

Secchi disk depths were generally smallest (turbidity highest) in July at all four locations (Fig. 5). Lowest turbidity (deepest secchi disk depths) generally occurred at Waretown, highest turbidity at Manahawkin, and intermediate turbidity at Mantoloking and Holly Park.

Phytoplankton primary production rates (gross) measured at near saturated light intensities (Fig. 6a) did not show as strong a seasonal pattern as chlorophyll *a* concentrations (Fig. 4). Rates were generally highest at Manahawkin where chlorophyll *a* concentrations were often highest; rates were generally lowest at Waretown where chlorophyll *a* concentrations were often lowest.

Light intensity is an important variable affecting photosynthesis rates in estuaries (Parsons et al. 1977). Rates of daily *in situ* phytoplankton production rates were calculated according to the algorithm developed by Keefe et al. (1981) for each station in Barnegat Bay using secchi disk depths and photosynthesis rates measured at saturated light intensity. Rates of *in situ* daily phytoplankton production were similar at all four stations in Barnegat Bay (Fig. 6b). During one sampling date (August) there was some indication that rates were higher at Manahawkin, relative to the other three stations.

A more accurate calculation of *in situ* phytoplankton production rates requires direct measurements of photosynthesis as a function of light intensity. While such measurements were not made by NJDEPE during 1989, they were made during the 1990 field season. Those data will be used to calculate 1990 phytoplankton production rates in a later report, and will provide additional insight into the accuracy of using the Keefe et al. (1981) algorithm in Barnegat Bay.

Sediment-Water Nutrient and Oxygen Fluxes

Sediment-water fluxes of oxygen, measured as a function of light intensity, demonstrated that the sediments in Barnegat Bay are active sites for decomposition of organic matter and, at times, active sites for benthic photosynthesis (Table 1; Appendix A, Figs. A-1A to A-6A). Rates of oxygen consumption in the dark reflect benthic metabolism (organic matter decomposition) (Eq. 5). Rates of benthic metabolism were highest in June, at both the sandy and silt-clay sites (Table 1; Figs. 7-8). Rates were approximately twice as high at the sandy site relative to the silt-clay site in June (Fig. 9). Rates decreased by August and were similar at both sites; rates in October were similar to those in August.

Gross (total) benthic photosynthesis is the sum of net photosynthesis (oxygen flux in the light) plus the rate of benthic respiration (determined from rates of dark oxygen consumption) (Eqs. 5-7). Gross benthic photosynthesis rates at ambient mid-day bottom light intensities (150 μE m^{-2} sec^{-1}) were high (6675 μmol O m^{-2} h^{-1}) at the sandy site in June; rates were greatly reduced in August and increased in October (3090 μmol O m^{-2} h^{-1}) to approximately half those measured in June (Table 1; Fig. 10). At the silt-clay site, rates of gross benthic photosynthesis were low during all three sampling periods (range approximately 150 to 1085 μmol O m^{-2} h^{-1}) (Table 1), as were light intensities ($\leq$20 μE m^{-2} sec^{-1}) (Table 1).

Benthic fluxes of phosphate were not statistically different from 0 (ρ=0.05) at either the sandy or silt-clay site during any of the three sampling periods, under any light regime (Table 1; Appendix A, Figs. A-1B to A-6B). However, based on the amount of organic matter decomposed in the sediments, calculated from O_2 consumption rates (Eq. 5) and Redfield stoichiometry (Eq. 1), a considerable amount of phosphate should have been released (Table 1; Appendix A, Figs. A-1B to A-6B). Predicted rates of phosphate release from the sediments in the dark ranged from 7 to 30 μmol P m^{-2} h^{-1} (Table 1).

Benthic fluxes of nitrate generally were not statistically different from 0 ($\rho=0.05$) at either sampling location (Table 1; Appendix A, Figs. A-1D to A-6D). Small fluxes ($\leq \pm$ 10 μmol m^{-2} h^{-1}) of nitrate were noted on only two occasions at each site.

Ammonia fluxes were $\leq$20 μmol m^{-2} h^{-1} at the sandy site in June and October; in August there was a considerable flux of ammonia out of the sediments ranging from 70 to 150 μmol N m^{-2} h^{-1} (Table 1; Appendix A, Figs. A-1C, A-3C and A-5C). At the silt-clay site, there was a considerable release of ammonia in June and August and no measurable flux in October (Table 1; Appendix A, Figs. A-2C, A-4c and A-6C).

A comparison of laboratory and *in situ* measured fluxes of nutrients and oxygen under dark and light conditions at the sandy vegetated site in June demonstrated that fluxes measured in the field and laboratory are similar. Both laboratory and *in situ* ammonium data demonstrated no release of ammonium from the sediments (Appendix A, Figs. A-1C and A-7C). Similarly, both laboratory and *in situ* nitrate and phosphate fluxes were negligible (Appendix A, Figs. A-1B, A-1D, A-7B and A-7D).

Oxygen fluxes in the light were also similar in the water overlying both the field and laboratory incubated sediments (Appendix A, Figs. A-1A and A-7A). Measurements of oxygen consumption in the dark indicate that rates were similar although somewhat higher in the laboratory than in the field (Appendix A, Figs. A-1A and A-7A). One of the differences between the field and laboratory techniques is the use of the benthic flux chamber for the *in situ* fluxes. The chamber has a thin plastic disposable glove attached to the side to allow outside water to fill the space inside the chamber when samples are withdrawn without contaminating the chamber water. Oxygen could diffuse across the glove resulting in lower apparent field oxygen consumption rates. In addition, there is some leakage of outside water into the *in situ* chambers when they are stirred before each oxygen sample, as the stopper holding the stirrer must be loosened to turn the stirrer.

Water Column Enclosures and Sediment Deposition Rates

Conditions inside and outside the enclosures were demonstrated to be similar based on a variety of measurements. Profiles of light intensity were similar inside and outside the enclosures (Fig. 11). Phytoplankton species composition and abundances (Table 2) and phytoplankton production rates inside and outside the enclosures (Table 3) were similar, measured 48 h after enclosure deployment. In addition, there were no measurable differences in ammonium, nitrate and phosphate concentrations inside and outside the enclosures (Table 4). The results described above indicate that conditions inside and outside the enclosures were similar.

During the previous year's study, the water column enclosures were subject to heavy weather and vandalism; unfortunately, 1989 was no exception. Some or all of the

enclosures deployed on 1, 8 and 11 August, and 14 and 18 September were lost to heavy weather or vandalism (Table 5).

There were measurable rates of particulate P, N and C deposition at the two study sites on all dates examined (Table 5; Figs. 12-14). At the sandy vegetated site, rates of P, N and C deposition appeared higher in early August (as well as in early July when only P data are available) compared to the early to mid-September measurements; however, because of the small number of sampling dates, we cannot reliably infer that this apparent trend was real. Measurements of particulate P, N and C deposition are only available for September at the silt-clay site because of loss of enclosures in August due to heavy weather or vandalism (Table 5). Rates of P, N and C deposition at the silt-clay site on 1 and 8 September were similar to those measured at the sandy vegetated site in early September (Table 5), as would be expected since the two sites are within approximately 1 km of each other and the water is likely well mixed between the sites. At both sites, it appears that rates of particulate P, N and C deposition were higher on 29 September relative to the early September measurements. The average ($\pm$S.D.) P, N and C deposition rates measured in this study at the sandy vegetated site are 24 (14) μmol P m^{-2} h^{-1}, 462 (146) μmol N m^{-2} h^{-1}, and 5113 (1374) μmol C m^{-2} h^{-1} (Table 5). At the silt-clay site average ($\pm$S.D.) deposition rates are 30 (29) μmol P m^{-2} h^{-1}, 485 (226) μmol N m^{-2} h^{-1}, and 5347 (2261) μmol C m^{-2} h^{-1} (Table 5). The C deposition rates are approximately 80% of the average summer phytoplankton production rate (6740 μmol C $m^{-2}h^{-1}$).

The ratios of N:P, C:N and C:P of the sediment trap material were not statistically different at the two sites (Table 5). The C:P ratio (392:1) was almost four times greater than the theoretical ratio of 106:1. The C:N ratio (aver. 11$\pm$1) was approximately two times greater than the theoretical ratio of 6.625:1, and the N:P ratio averaged 33:1, which is approximately two times greater than the theoretical Redfield ratio of 16:1. Together, these data indicate that the organic matter deposited to the sediments from the water column in Barnegat Bay is not typical "Redfield type" organic matter, but rather is depleted in both nitrogen and phosphorus relative to carbon.

N and P Burial in Sediments

The P and N contents of the sediments at the sandy vegetated site showed no consistent increase or decrease with depth (Figs. 15 and 16). Total P concentrations ranged from approximately 0.1 to 0.24 mg P/gds (gram dry sediment). Approximately 90% or more of the total P was in the form of inorganic P, except in the top 1 cm where inorganic P accounted for approximately 70% of the total P (Fig. 15). Organic P generally accounted for less than 10% of total P, except in the top 1 cm where it accounted for approximately 30% of the total P content (Fig. 15); this likely reflects recently deposited organic matter which has not yet been metabolized. (At times the inorganic P content appears slightly higher than the total P which is due to analytical variability.) In contrast to P, over 90% of the total N in the sediments at the sandy vegetated site was in the form of organic N, with less than 10% as inorganic extractable ammonia (Fig. 16). Concentrations of total N ranged from approximately 0.2 mg

N/gds to 0.9 mg N/gds. The total N content increased approximately 2 fold between 3 and 5 cm, and the total P increased slightly. We have no direct evidence to explain this; it may be due to increased inputs from the roots and rhizomes of seagrasses.

At the silt-clay site profiles of both P and N content with depth gradually decreased from the surface down to approximately 4 to 5 cm (Figs. 17 and 18). Total P concentrations were approximately twice as high as at the sandy site and ranged from approximately 0.3 to 0.45 mg P/gds with 80% or more of the P in the form of inorganic P. Total N concentrations were two to three times greater than at the sandy site and ranged from approximately 0.8 to 1.4 mg N/gds. Extractable ammonia concentrations were highest at the surface (0.06 mg N/gds) and decreased rapidly with depth to less than 0.007 mg N/gds (Fig. 18), and were always less than 10% of organic N.

At the silt-clay site the profile of ^{137}Cs showed a peak between 0 and 4 cm (R. Bopp unpubl. data). Based on the ^{137}Cs data, maximum net sediment accumulation rates were estimated to be approximately 0.1 cm/y (R. Bopp, pers. comm.). In the surface 0-2 cm ^{7}Be was measured as 679 $\pm$99 dpm with activity decreasing to 23 $\pm$75 dpm at 2-4 cm (R. Bopp, unpubl. data). Radioisotope profiles were not measured on cores from the sandy site.

Based on a net sediment accumulation rate at the silt-clay site of 0.11 cm/y, an average measured sediment density of 1.5 gds/cm^3 wet sediment, and the average total N and P content in the top 4 cm of sediment, rates of net N and P burial were calculated to be 135 mmol N m^{-2} y^{-1} and 18 mmol P m^{-2} y^{-1}, respectively, at the silt-clay site.

DISCUSSION

The major objectives of the current study were to: 1) assess the temporal and spatial variability of water quality in Barnegat Bay, and 2) begin to quantify the important processes that control nutrient availability and thus eutrophication within the Bay, including the magnitude of the sediments as a source or removal site for nutrients.

Temporal and Spatial Variability of Water Quality

Chlorophyll concentrations, phytoplankton production rates and water column turbidities indicate that Barnegat Bay is currently in a moderately eutrophic state. Phytoplankton production rates, measured between June and October, are markedly higher than rates in many other East Coast estuaries, including Narragansett Bay, Delaware Bay and Charleston Pond (a Rhode Island coastal lagoon), Chesapeake Bay, and Great South Bay, LI, also a coastal lagoon (Fig. 19). All of these estuaries receive substantial amounts of N and P loading from anthropogenic sources.

Chlorophyll concentrations are also high in Barnegat Bay compared to other estuaries with high nutrient loading rates and are similar to Great South Bay, LI (Fig. 20). Light attenuation coefficients, and thus water column turbidities, are higher in Barnegat Bay than many other coastal lagoons (Fig. 21). Two factors that contribute to water column turbidity are phytoplankton biomass and suspended sediment. Both are likely important in Barnegat Bay. The high phytoplankton production rates and high chlorophyll concentrations indicate high phytoplankton biomass in the water column which absorbs light and decreases light penetration. In addition, because of the shallow water column, winds and probably heavy boating activity result in considerable resuspension of bottom sediments into the water. While currently there is sufficient light for the phytoplankton to be able to use essentially all of the nutrients available in the water column, the high water column turbidity has important implications for the continued occurrence of seagrasses and benthic algae in the Bay.

Currently, little light is reaching the bottom of Barnegat Bay in the mid- to late summer, even in areas only 1 to 1.5 m deep (Table 1). These low light levels result in very low rates of benthic primary production by seagrasses and benthic algae in mid- to late summer, even at the shallow sandy study site, and low rates throughout the summer at the deeper silt-clay study site (Table 1; Fig. 10). In fact, while *Zostera*

was abundant in June at our vegetated study site, no *Zostera* was growing at that location later in the summer, and by August it was absent. The large decrease in rates of benthic photosynthesis between June and August at the sandy vegetated site, may in part reflect the low abundance of *Zostera* and/or *Ruppia* (<10% cover) in August, compared to their abundance (~25% cover) in June. Increased water column turbidity is considered to be a major factor leading to dramatic decreases in seagrass beds in other estuaries including Chesapeake Bay (Bayley et al. 1978; Orth and Moore 1983). These seagrass beds are an important habitat and nursery ground for finfish and shellfish.

Rates of benthic primary production at both the sandy and silt-clay sites were considerably less than rates of phytoplankton production in Barnegat Bay. Daily benthic photosynthesis rates were estimated based on the relationship between light intensity and benthic primary production (Table 1; Fig. 22; Eq. 2). Highest rates of daily benthic primary production occurred in June at the sandy site (approximately 30 mmol C $m^{-2}d^{-1}$), which is approximately four times less than phytoplankton primary production rates in the Bay in June (140 mmol C $m^{-2}d^{-1}$; Fig. 6b). In October, rates of daily benthic primary production at the sandy site were approximately 10 mmol C $m^{-2}d^{-1}$ compared to phytoplankton primary production rates of approximately 100 mmol C $m^{-2}d^{-1}$. Rates of benthic primary production may be higher at other locations in Barnegat Bay where there is a higher biomass of SAVs. While our methodology did not permit us to distinguish benthic primary production due to SAVs versus that due to benthic algae, highest benthic primary production rates occurred when SAV abundance (approximately 25% cover) was greatest (June at sandy site).

Factors Controlling Nutrient Availability and Eutrophication

Sources of nutrients supporting phytoplankton production and thus eutrophication in estuaries include external inputs (e.g., rivers, runoff, groundwater) and internal recycling in the water column and in benthic sediments. The flux of inorganic nutrients from sediments to the water column following decomposition of organic matter in the sediments often supplies between 25% and 50% or more of the nitrogen and phosphorus requirements of phytoplankton (Nixon 1981; Kemp et al. 1982; Boynton and Kemp 1985). However, measurements of benthic nutrient fluxes have been made primarily in estuaries in which the average water depth is greater than the 1% light compensation depth resulting in no or low benthic algal and seagrass primary production (Table 6). Few measurements of benthic nutrient fluxes have been made in shallow coastal lagoons where there is sufficient light penetration for substantial benthic algal production (Nowicki and Nixon 1985a,b). In such systems, all or some of the nutrients mineralized in the sediments may be assimilated at the sediment surface by benthic algae.

Based on the rates of benthic metabolism measured in Barnegat Bay sediments and a Redfield stoichiometry for organic matter decomposition (Eq. 1), considerable

amounts of both N and P should be released from the sediments (Table 1). However, none of the P and only some of the N released during the decomposition of organic matter in the Bay sediments is recycled to the water column by diffusive flux. The amount of N and P that appears not to be returned to the water column is equivalent to between 20% and 50% of the estimated phytoplankton N and P requirements.

In Barnegat Bay, NJ, benthic algal production is important in controlling the flux of nutrients, particularly nitrogen, out of the sediments. There was no measurable flux of ammonia out of the sediments at either the sandy or silt-clay site when gross daytime benthic photosynthesis rates were approximately 900 μmol O m^{-2} h^{-1} or greater (Fig. 23). Even when cores which had benthic photosynthesis rates $\geq$900 μmol O m^{-2} h^{-1} in the light were incubated in the dark for periods of 6 to 24 h, no ammonia was released (Table 1). However, when ambient field benthic photosynthesis rates were low (approximately 450 μmol m^{-2} h^{-1} or less; June silt-clay site and August sandy and silt clay sites), the flux of ammonia out of the sediments was similar to that predicted from Redfield stoichiometry (Fig. 23; Table 1). Light intensity is an important factor controlling rates of benthic photosynthesis in Barnegat Bay (and thus the amount of ammonia recycled from the sediments to the overlying water), based on the linear relationship between light intensity and benthic photosynthesis (Fig. 22). Low light levels at the sediment surface during mid-summer (July-August) resulted in low rates of benthic primary production, while higher rates of benthic primary production were measured in early summer (June) and fall (October) when bottom light levels were higher. Submerged aquatic vegetation (SAV) does not appear to be a major factor controlling sediment-water nutrient fluxes at our study sites, as patterns of N and P release were similar regardless of the presence or absence of SAVs. Further experiments confirming the importance of benthic algae in controlling sediment-water ammonia fluxes were carried out during YR III and will be included in the YR III Final Report.

Durand (1984) found significant release of NH_4 from coastal lagoon sediments in Absecon Bay, NJ. However, it is difficult to compare his results directly with our Barnegat Bay data because Durand did not report ambient bottom light intensities and measurements were made in the dark.

The lack of measurable sediment-water fluxes of nitrate at either station in Barnegat Bay at any time suggests that sediment nitrification rates were low. It also could be due to very efficient denitrification of any nitrate produced in the sediments (Seitzinger 1987b). Denitrification can be a significant sink for nitrogen in estuaries, removing as much as 50% of the external N loading (Seitzinger 1988). Measurements of denitrification were not made in the current study. In addition to denitrification, permanent burial in the sediments can be a sink for N in estuaries. However, rates of N burial in the sediments were small (135 mmol N m^{-2} y^{-1} translated into units of μmol m^{-2} h^{-1} for comparison with benthic nutrient flux data is 15 μmol N m^{-2} h^{-1}) relative to the difference between the measured and predicted release of N from the sediments (Table 1).

While benthic algae appear to be important in controlling sediment-water ammonia fluxes, the data indicate that factors in addition to benthic algae control phosphate fluxes. There was no measurable release of phosphate from the sediments at either the sandy or silt-clay site regardless of whether benthic algal production rates were high or low, or whether sediments were incubated in the light or the dark (Table 1). Burial of iron phosphate complexes is an important mechanism of phosphate retention in freshwater sediments (Wetzel 1985). However, the inorganic phosphorus content of sediments at both the sandy and silt-clay sites was relatively low (Figs. 15 and 17) and net burial of P in the sediments was small (18 mmol P m^{-2} y^{-1} = 2 μmol P m^{-2} h^{-1}) relative to the difference between the measured and predicted release of P from the sediments. More extensive investigations of the mechanisms involved in sediment-water phosphate dynamics including phosphate sorption to sediments, resuspension of bottom sediments, such as may occur due to wave action or boating activity, the C:N:P ratio of decomposing organic matter, and benthic algal interactions, were conducted during YR III and will be included in the YR III Final Report.

Implications for Eutrophication in Barnegat Bay

Nutrient concentrations are currently relatively low in Barnegat Bay, in part, because there is sufficient light for the phytoplankton to assimilate the current rate of nutrient inputs. While phytoplankton production rates in Barnegat Bay are already considerably higher than in many estuaries, an increase in nutrient inputs to Barnegat Bay, from either external sources or from the sediments, would be expected to further increase phytoplankton production, and thus increase eutrophication. (Studies of factors controlling phytoplankton production in Barnegat Bay were not a part of the current study but were investigated with nutrient enrichment studies using mesocosms during YR IV. A strong response of phytoplankton production to increased nutrient loading was found; the results will be presented in full in the YR IV Final Report.)

An increase in external N and P inputs to the Bay would increase phytoplankton biomass in the water, increasing water column turbidity, thus decreasing light to the sediment surface. Currently, both N and P are being retained/removed efficiently in the sediments, thus decreasing the magnitude of eutrophication in the Bay at the present rate of external nutrient input. This appears to be, in part, due to benthic algae on the sediment surface which are assimilating the nutrients regenerated in the sediments. Light is a major limiting factor for benthic primary production in the Bay, as indicated by the close relationship between light intensity and benthic primary production (Fig. 10) and benthic primary production is important in controlling N (Fig. 22), and also likely P fluxes from the sediments. If water column turbidities increase to the point that benthic algal production is no longer sufficient to assimilate N and P regenerated in the sediments, then the flux of N and P from the sediments to the water column would be expected to increase and to follow patterns similar to those in deeper estuaries, where bottom light intensities are not sufficient for significant benthic algal production.

The more efficient removal of P relative to N in the sediments (as well as the overall high efficiency of removal of both N and P relative to deeper estuaries) demonstrates that nutrient processes in Barnegat Bay differ quite dramatically from nutrient processing in deeper estuaries. No P is released from the sediments in Barnegat Bay, while at times some N (as NH_4) is returned to the water column. Such a high efficiency of P or N removal in the sediments has never been reported in other estuaries. Evidence is beginning to accumulate from other shallow estuaries suggesting that sediments in coastal lagoons are more efficient at removing P than N (Seitzinger unpubl. data; Nowicki and Nixon 1985). This differs from deeper estuaries where N is generally removed more efficiently than P (Nixon 1981).

The findings of this study have important implications for management decisions regarding future control of nutrient inputs to Barnegat Bay, as models of nutrient control based on the relationship between nutrient inputs and eutrophication developed for deeper estuaries are not applicable to Barnegat Bay and likely to other shallow back bay estuaries. We are working currently with the U.S. Army Corps of Engineers (Waterways Experiment Station) in modifying the Chesapeake Bay 3-D hydrodynamic/water quality model for application to coastal lagoons, based on our data from Barnegat Bay, as well as data we are collecting in the Delaware Inland Bays.

LITERATURE CITED

American Public Health Association, American Water Works Association and Water Pollution Control Federation (APHA, AWWA and WPCF). 1989. Standard methods for the examination of water and wastewater, 17th ed. Washington, D.C.

Bayley, S., V.D. Stotts, P.F. Springer and J. Steenis. 1978. Changes in submerged aquatic macrophyte populations at the head of Chesapeake Bay, 1958-1975. Estuaries 1:73-84.

Bloesch, J. and N.M. Burns. 1980. A critical review of sedimentation trap technique. Schweiz. Z. Hydrol. 42:15-51.

Boynton, W.R. and W.M. Kemp. 1985. Nutrient regeneration and oxygen consumption by sediments along an estuarine salinity gradient. Mar. Ecol. Prog. Ser. 23:45-55.

_____ and C.W. Keefe. 1982. A comparative analysis of nutrients and other factors influencing estuarine phytoplankton production. Pages 69-90 *in* V.S. Kennedy, ed. Estuarine Comparisons. Academic Press, NY. 709 pp.

_____ and C.G. Osborne. 1980. Nutrient fluxes across the sediment-water interface in the turbid zone of a coastal plain estuary. Pages 93-109 *in* V.S. Kennedy, ed. Estuarine Perspectives. Academic Press.

Carpenter, J.H. 1965. The Chesapeake Bay Institute technique for the Winkler dissolved oxygen method. Limnol. Oceanogr. 10:141-143.

Culberson, C. M., J.R. Pennock, B.W. Lee, R.B. Biggs, T.M. Church and J.H. Sharp. 1987. Data from the YABLED Cruises, September 1981-July 1984. U. Delaware Oceanographic Data Rept. No. 4. Delaware Sea Grant Coll. Prog., U. Delaware.170 pp.

D'Elia, C.F., J.G. Sanders and W.R. Boynton. 1986. Nutrient enrichment studies in a coastal plain estuary: Phytoplankton growth in large-scale, continuous cultures. Can. J. Fish. Aquat. Sci. 43:397-406.

Durand, J.B. 1984. Nitrogen distribution in New Jersey coastal bays. Pages 29-51 *in* M.J. Kennish and R.A. Lutz, eds. Lecture notes on coastal and estuarine studies. 6. Ecology of Barnegat Bay, New Jersey. Springer-Verlag, NY. 396 pp.

Fisher, T.R., P.R. Carlson and R.T. Barber. 1982. Sediment nutrient regeneration in three North Carolina estuaries. Est. Coast. Shelf Sci. 14:101-116.

Froelich, P.N., M.A. Arthur, W.C. Burnett, M. Deakin, V. Hensley, R. Jahnke, L. Kaul, K.-H. Kim, K. Roe, A. Soutar and C. Vathakanon. 1988. Early diagenesis of organic matter in Peru continental margin sediments: Phosphorite precipitation. Mar. Geo. 80:309-343.

Gardner, W.D. 1980. Field assessment of sediment traps. J. Mar. Res. 38:41-52.

Harding, L.W., Jr., B.W. Meeson and T.R. Fisher, Jr. 1986. Phytoplankton production in two East Coast estuaries: Photosynthesis—light functions and patterns of carbon assimilation in Chesapeake and Delaware bays. Estuarine, Coastal and Shelf Sci. 23:773-806.

Hargrave, B.T. and N.M. Burns. 1979. Assessment of sediment trap collection efficiency. Limnol. Oceanogr. 24:1124-1135.

Hargrave, B.T. and S. Taguchi. 1978. Origin of deposited material sedimented in a marine bay. J. Fish. Res. Bd. Can. 35:1604-1613.

Hartwig, E.O. 1976. The impact of nitrogen and phosphorus release from a siliceous sediment on the overlying water. Pages 103-117 *in* M. Wiuley, ed. Estuarine Processes, Vol. 1. Academic Press, NY.

Hopkinson, C.S. and R.L. Wetzel. 1982. *In situ* measurements of nutrient and oxygen fluxes in a coastal marine benthic community. Mar. Ecol. Prog. Ser. 10:29-35.

Keefe, C.W., W.R. Boynton and W.M. Kemp. 1981. A review of phytoplankton processes in estuarine environments. UMD CEES Ref. No. [UMCEES] CBL 81-193, Solomons, MD. 4 pp.

Kelly, J.R. and S.A. Levin. 1985. A comparison of aquatic and terrestrial nutrient cycling and production processes in natural ecosystems, with reference to ecological concepts of relevance to some waste disposal issues. *In* NATO Advanced Research Workshop. Scientific Basis for the Role of the Oceans as a Waste Disposal Option. Vilamoura, Portugal, April 1985, ERC-091. 29 pp.

Kemp, W.M., R.L. Wetzel, W.R. Boynton, C.F. D'Elia and J.C. Stevenson. 1982. Nitrogen cycling and estuarine interfaces: Some current concepts and research directions. Pages 209-230 *in* V.S. Kennedy, ed. Estuarine Comparisons. Academic Press, NY. 709 pp.

Lively, J.S., Z. Kaufman and E.J. Carpenter. 1983. Phytoplankton ecology of a barrier island estuary: Great South Bay, New York. Estuarine, Coastal & Shelf Sci. 16:51-68.

Martin, J.H. and G.A. Knauer. 1973. The elemental composition of plankton. Geochim. Cosmochim. Acta 37:1639-1653.

Murphy, J. and J.P. Riley. 1962. A modified single solution method for determination of phosphate in natural waters. Anal. Chim. Acta 27:31-36.

Nixon, S.W. 1981. Remineralization and nutrient cycling in coastal marine ecosystems. *In* B.J. Neilson and L.E. Cronin, eds. Estuaries and Nutrients. Humana Press, Clifton, NJ.

_____. 1983. Estuarine ecology—A comparative and experimental analysis using 14 estuaries and the MERL microcosms. Final Rept. to the USEPA, Chesapeake Bay Program. 59 pp.

_____. 1986. Nutrient dynamics and the productivity of marine coastal waters. Pages 97-115 *in* R. Halwagy, ed. Proceedings of the First Arabian Gulf Conference on Environment and Pollution. The Alden Press, Ltd., Oxford, England.

_____, J.R. Kelly, B.N. Furnas, C.A. Oviatt and S.S. Hale. 1980. Phosphorus regeneration and the metabolism of coastal marine bottom communities. Pages 219-242 *in* K.R. Tenore and B.C. Coull, eds. Marine Benthic Dynamics. Belle W. Baruch Library in Marine Sciences, No. 11.

_____ and V. Lee. 1981. The flux of carbon, nitrogen and phosphorus between coastal lagoons and offshore waters. Pages 325-348 *in* Coastal Lagoon Research, Present and Future. Div. of Marine Sciences, UNESCO, Paris.

_____, C.A. Oviatt and S.S. Hale. 1976. Nitrogen regeneration and the metabolism of coastal marine bottom communities. Pages 269-283 *in* J.M. Anderson and A. Macfadyen, eds. The Role of Terrestrial and Aquatic Organisms in Decomposition Processes. 17th Symposium, British Ecological Society. Blackwell Scientific Publications.

Nowicki, B.L. and S.W. Nixon. 1985a. Benthic community metabolism in a coastal lagoon ecosystem. Mar. Ecol. Prog. Ser. 22:21-30.

_____. 1985b. Benthic nutrient remineralization in a coastal lagoon ecosystem. Estuaries 8:182-190.

Orth, R.J. and K.A. Moore. 1983. Submerged aquatic vegetation of the Chesapeake Bay: Past, present and future. Science 222:51-53.

Oviatt, C.A. and S.W. Nixon. 1975. Sediment resuspension and deposition in Narragansett Bay. Est. Coast. Mar. Sci. 3:201-217.

Parsons, T.R., M. Takahashi and B. Hargrave. 1977. Biological Oceanographic Processes. 2nd ed. Pergamon Press. 332 pp.

Pennock, J.R. and J.H. Sharp. 1986. Phytoplankton production in the Delaware Estuary: Temporal and spatial variability. Mar. Ecol. Prog. Ser. 34:143-155.

Redfield, A.C. 1934. On the proportions of organic derivatives in sea water and their relation to the composition of plankton. Pages 176-192 *in* James Johnstone Memorial Volume. Liverpool Univ.

_____. 1958. The biological control of chemical factors in the environment. Am. Scientist 46:206-221.

Richards, F. 1965. Anoxic basins and fjords. Pages 611-645 *in* J.P. Riley and G. Skirrow, eds. Chemical Oceanography. Vol. 1, Chap. 13. Academic Press.

Seitzinger, S.P. 1987a. The effect of pH on the release of phosphorus from Potomac River sediments. USEPA Chesapeake Bay Program. CBP/TRS 15. November 1987. 50 pp.

_____. 1987b. Nitrogen biogeochemistry in an unpolluted estuary: The importance of benthic denitrification. Mar. Ecol. Prog. Ser. 41:177-186.

_____ and I.E. Pilling. 1990. Eutrophication and nutrient loading in Barnegat Bay: Initial studies of the importance of sediment-water nutrient interactions. Report No. 90-14. Acad. Nat. Sci. Phila. 50 pp.

Sellner, K.G., R.V. Lacouture, K.L. Heck, Jr., R.J. Orth and K.A. Moore. 1988. Phytoplankton, nutrients, macroalgae and submerged aquatic vegetation in Delaware's Inland Bays, 1985-1986. Rept. for Delaware Dept. Natural Res. and Environ. Control. Acad. Natural Sci. Phila. 140 pp.

Solorzano, L. 1969. Determination of ammonia in natural waters by the phenylhypochlorite method. Limnol. Oceanogr. 14:799-801.

Teague, K.G., C.J. Madden and J.W. Day, Jr. 1988. Sediment-water oxygen and nutrient fluxes in a river-dominated estuary. Estuaries 11:1-9.

Technicon Industrial Systems. 1977. Nitrate and nitrite in water and seawater. Industrial Method No. 158-71W/A. Technicon Industrial Systems, Tarrytown, NY. 4 pp.

Thayer, G.W. 1971. Phytoplankton production and the distribution of nutrients in a shallow unstratified estuarine system near Beaufort, N.C. Ches. Sci. 12:240-253.

Ullman, W.J. and M.W. Sandstrom. 1987. Dissolved nutrient fluxes from the nearshore sediments of Bowling Green Bay, Central Great Barrier Reef Lagoon (Australia). Estuarnie, Coastal and Shelf Sci. 24:289-303.

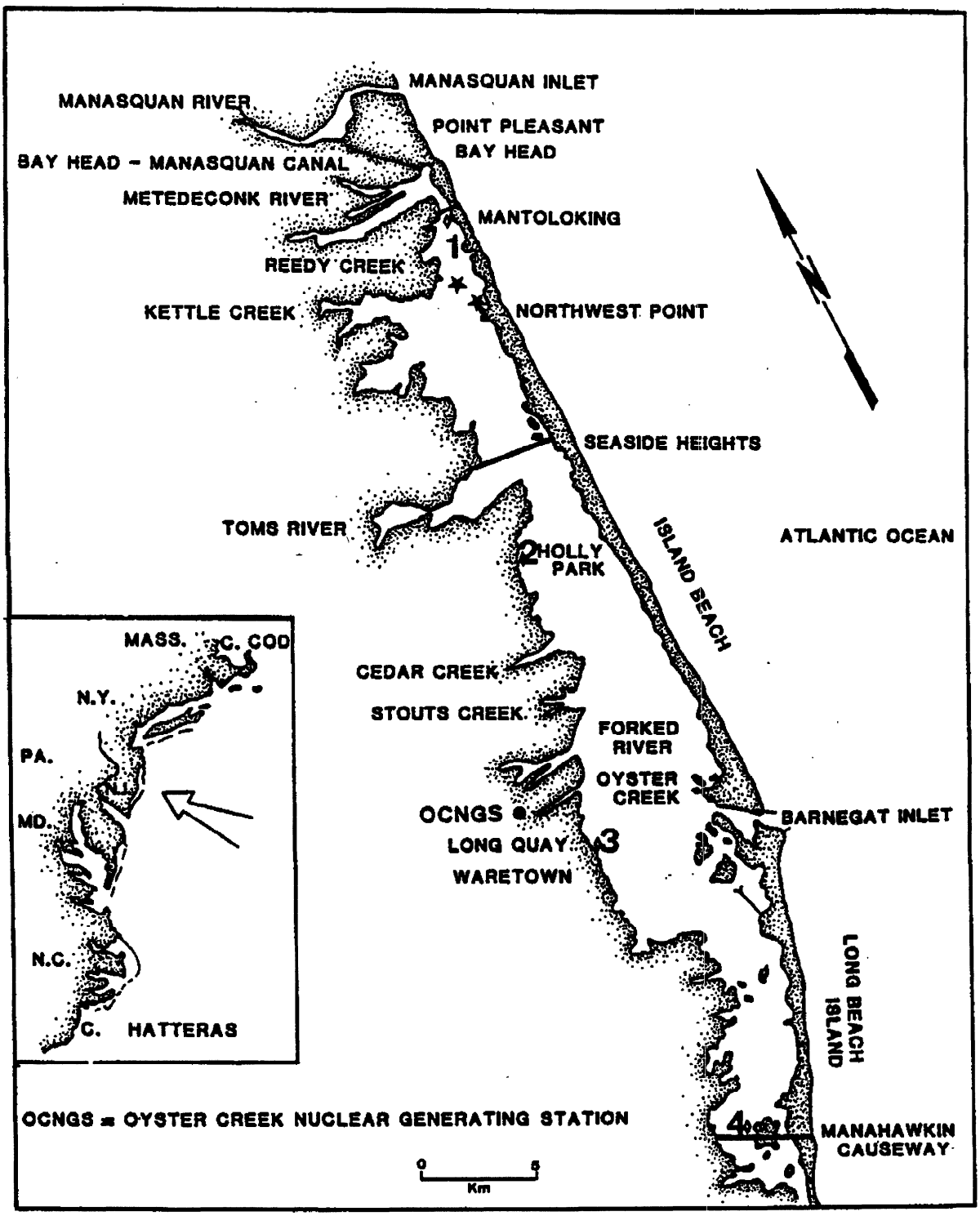


Figure 1. Map of Barnegat Bay, NJ, indicating NJDEPE DSR water column nutrient and primary production sampling stations (1-4) and ANSP sediment deposition and benthic flux stations (*). 1-Mantoloking; 2-Holly Park; 3-Waretown; 4-Manahawkin.

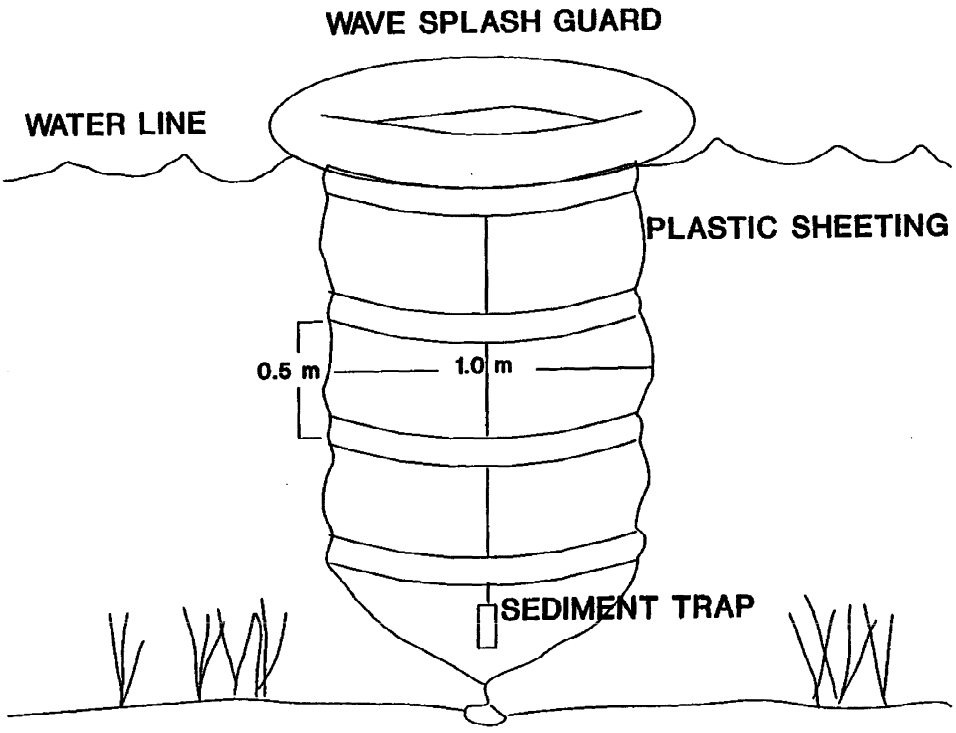


Figure 2. Water column enclosure with sediment trap for organic matter deposition measurements. See text for complete description of construction and use.

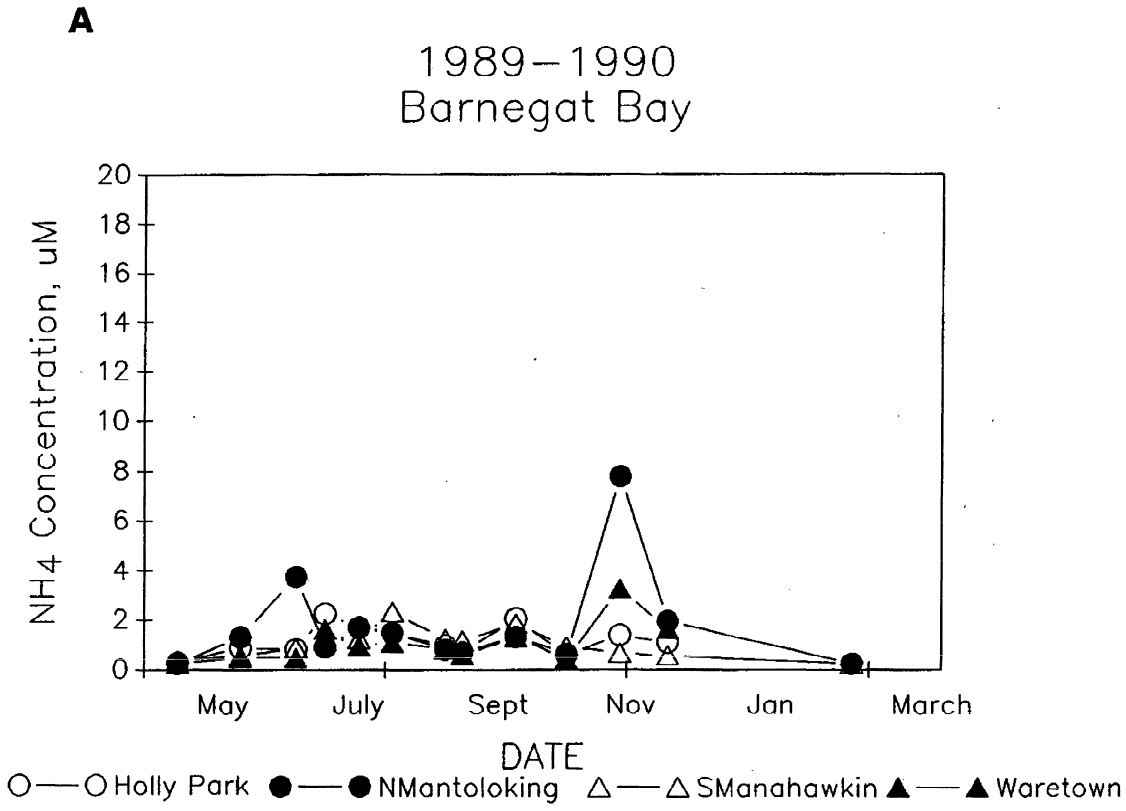


Figure 3. Nutrient concentrations, μM, at four locations in Barnegat Bay; A) ammonia, B) nitrate plus nitrite, and C) phosphate. See Figure 1 for station locations.

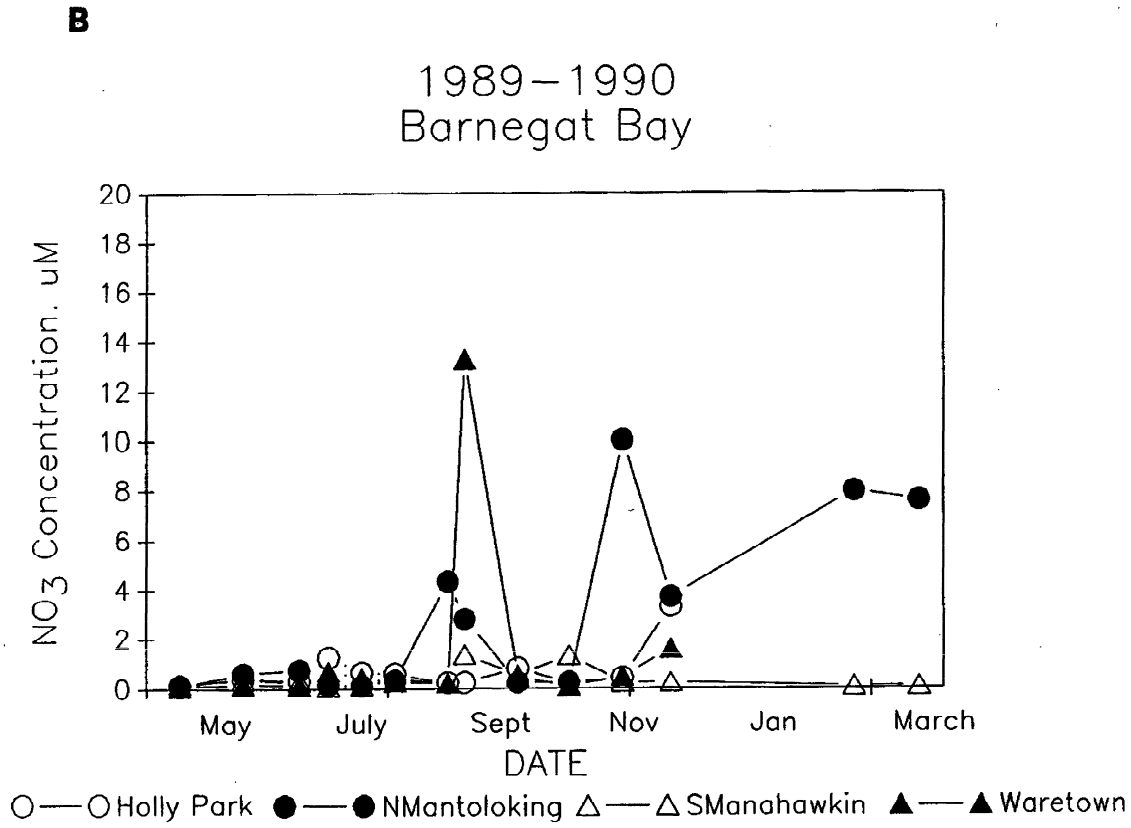


Figure 3 (continued). Nutrient concentrations, μM, at four locations in Barnegat Bay; A) ammonia, B) nitrate plus nitrite, and C) phosphate. See Figure 1 for station locations.

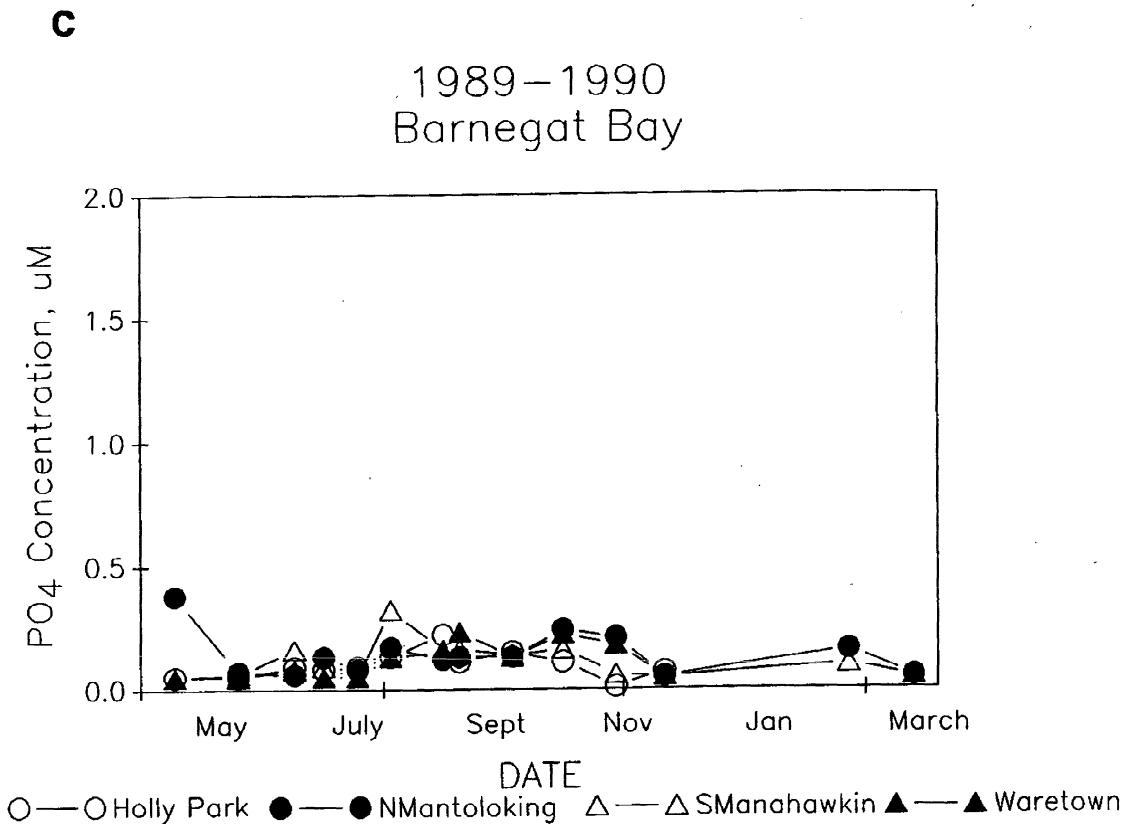


Figure 3 (continued). Nutrient concentrations, μM, at four locations in Barnegat Bay; A) ammonia, B) nitrate plus nitrite, and C) phosphate. See Figure 1 for station locations.

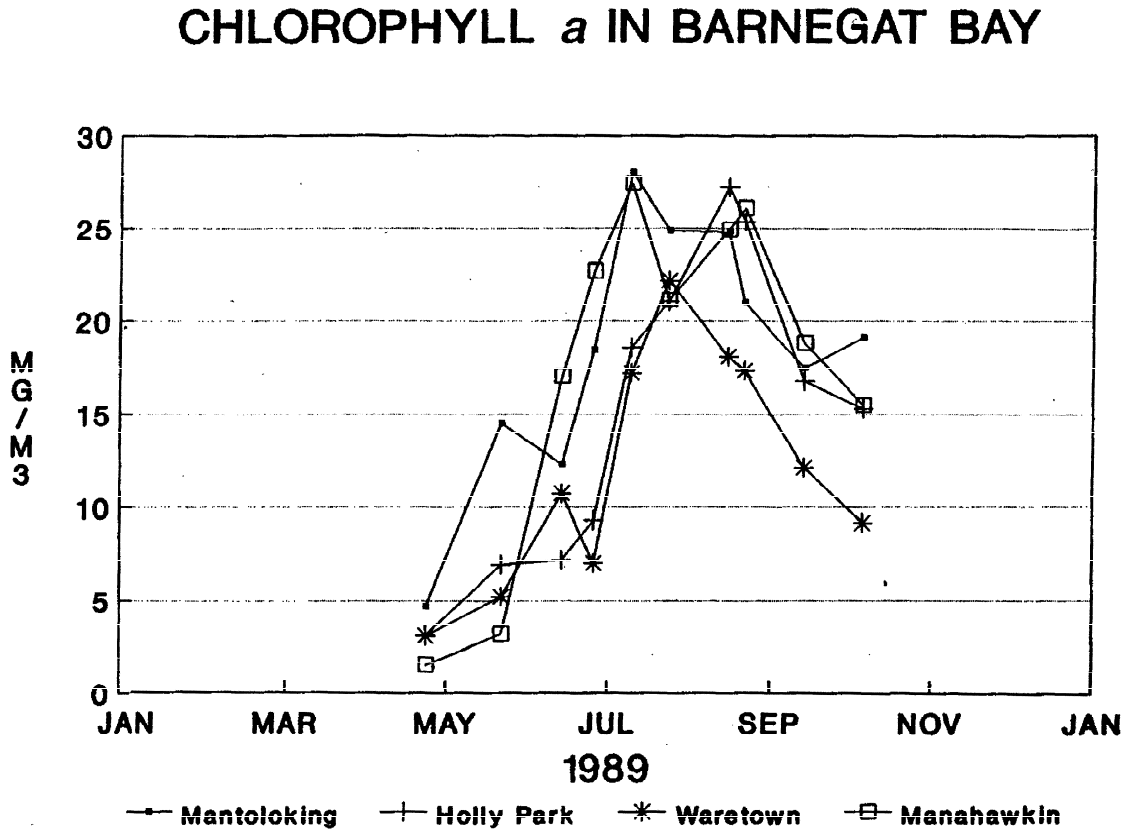


Figure 4. Chlorophyll *a* concentrations, mg/m^3, at four locations in Barnegat Bay, between April and October 1989. Data from NJDEPE Leeds Point Laboratory, unpubl. data.

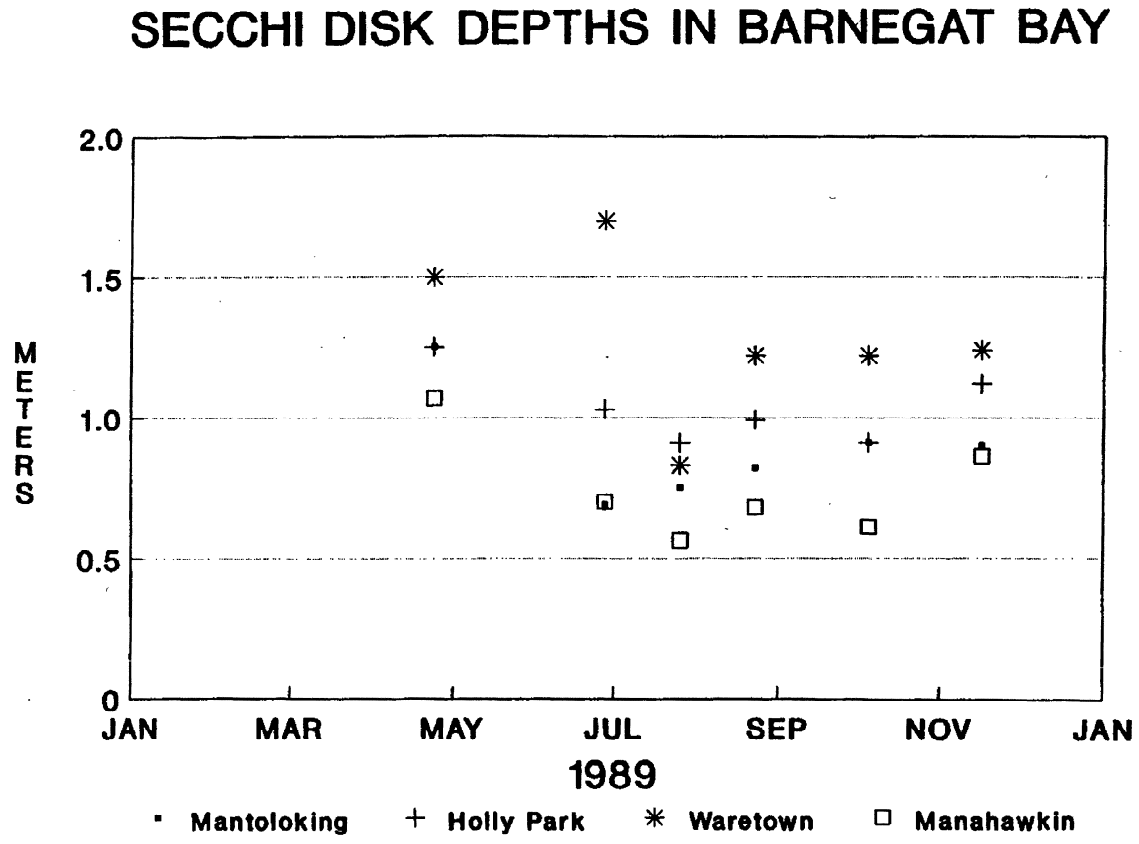


Figure 5. Secchi disk depths (m) at four locations in Barnegat Bay, between April and November 1989 (unpubl. data NJDEPE DSR).

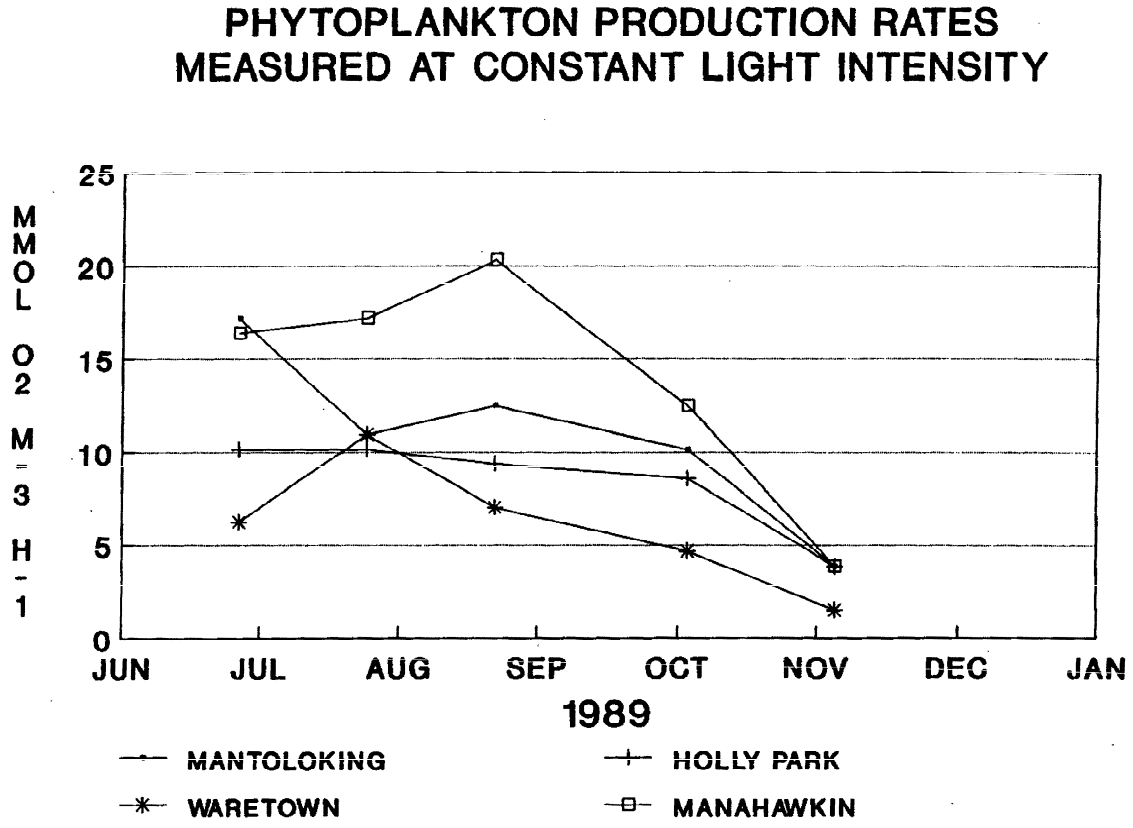


Figure 6A. Phytoplankton production rates (mid-day gross primary production) at four locations in Barnegat Bay, measured at constant light intensity between April and October 1989 (NJDEPE unpubl. data).

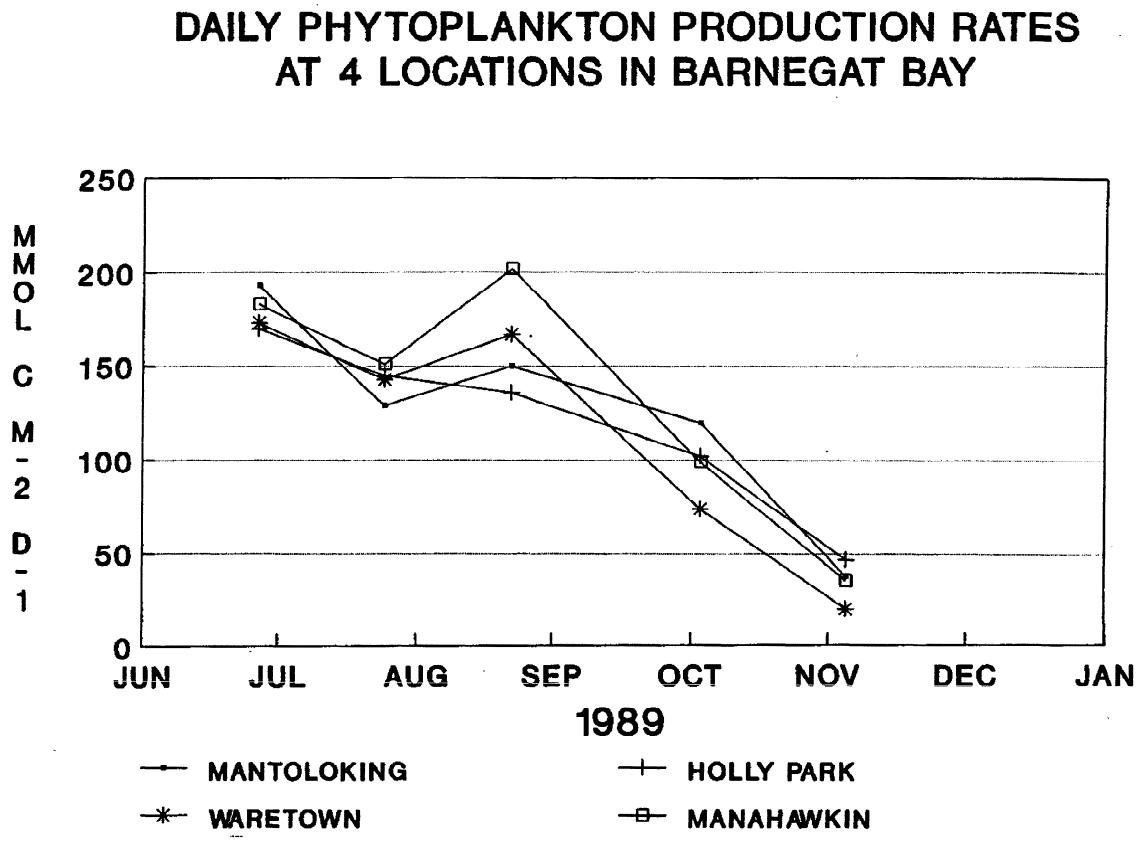


Figure 6B. Daily phytoplankton production rates at four locations in Barnegat Bay, calculated according to Keefe et al. (1981) from secchi disk depths (Fig. 5) and primary production rates at saturated light intensity (Fig. 6A).

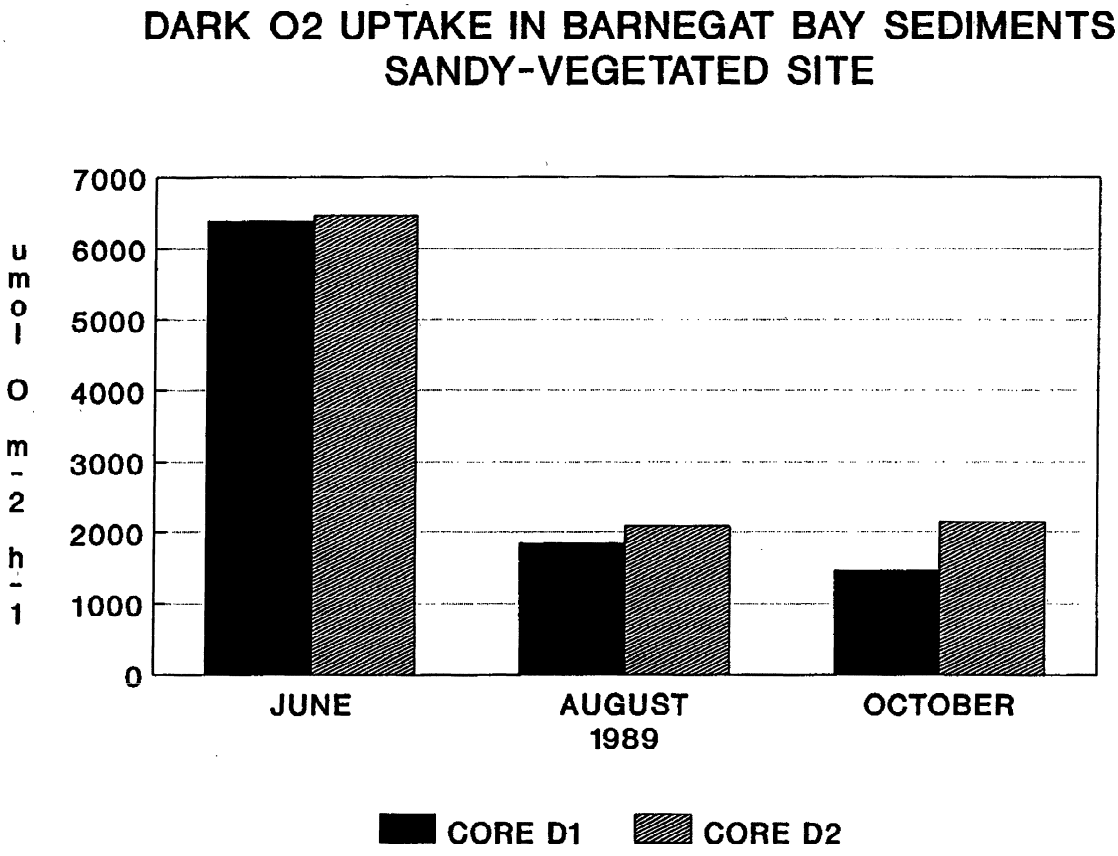


Figure 7. Rates of benthic metabolism (oxygen consumption rates in dark incubated cores) in duplicate cores collected from the sandy vegetated site in Barnegat Bay in June, August and October 1989.

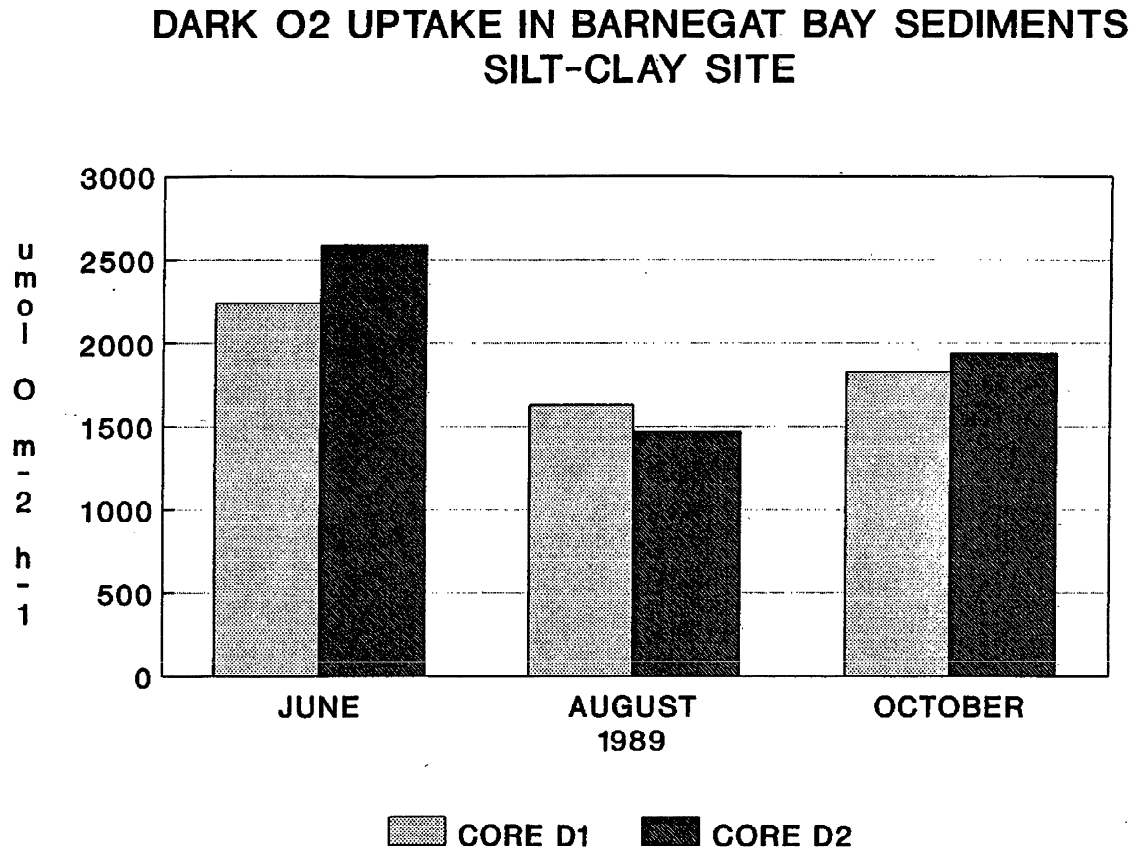


Figure 8. Rates of benthic metabolism (oxygen consumption rates in dark incubated cores) in duplicate cores collected from the silt-clay site in Barnegat Bay in June, August and October 1989.

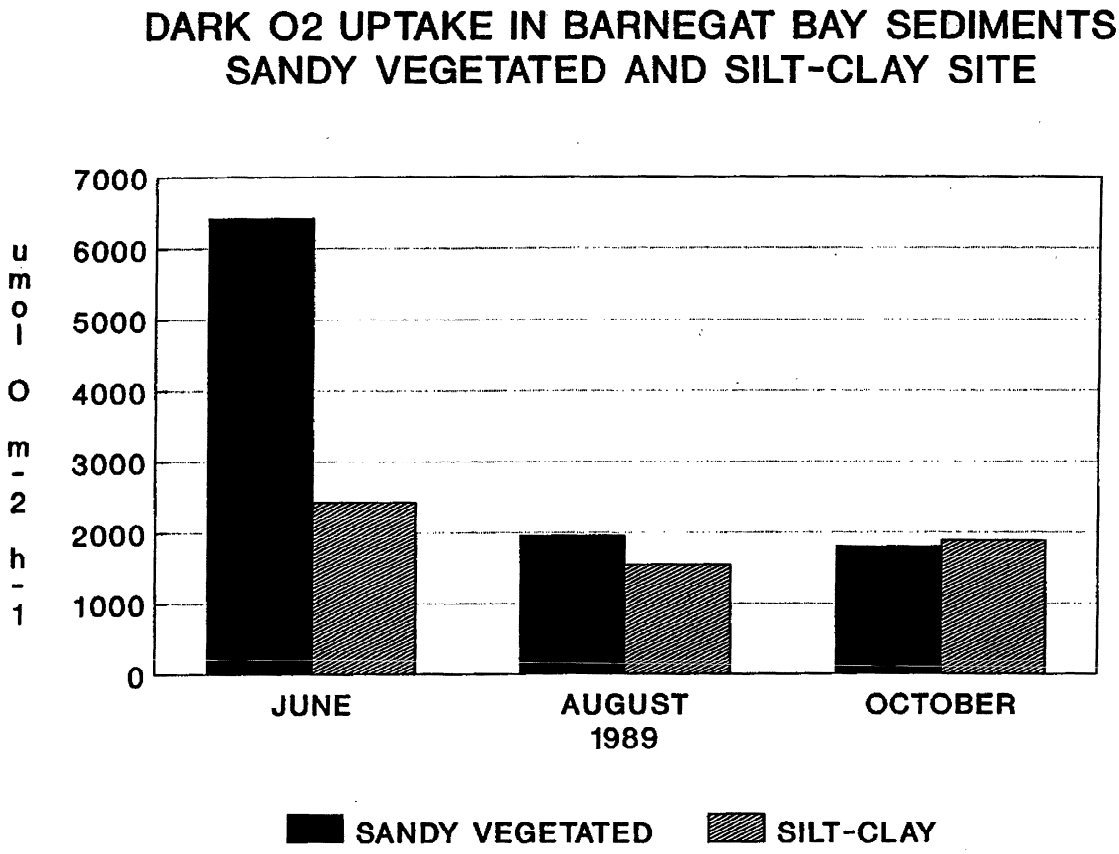


Figure 9. Comparison of the rate of benthic metabolism (average rate of oxygen consumption in duplicate dark incubated cores) at the sandy vegetated and silt-clay site in Barnegat Bay in June, August and October

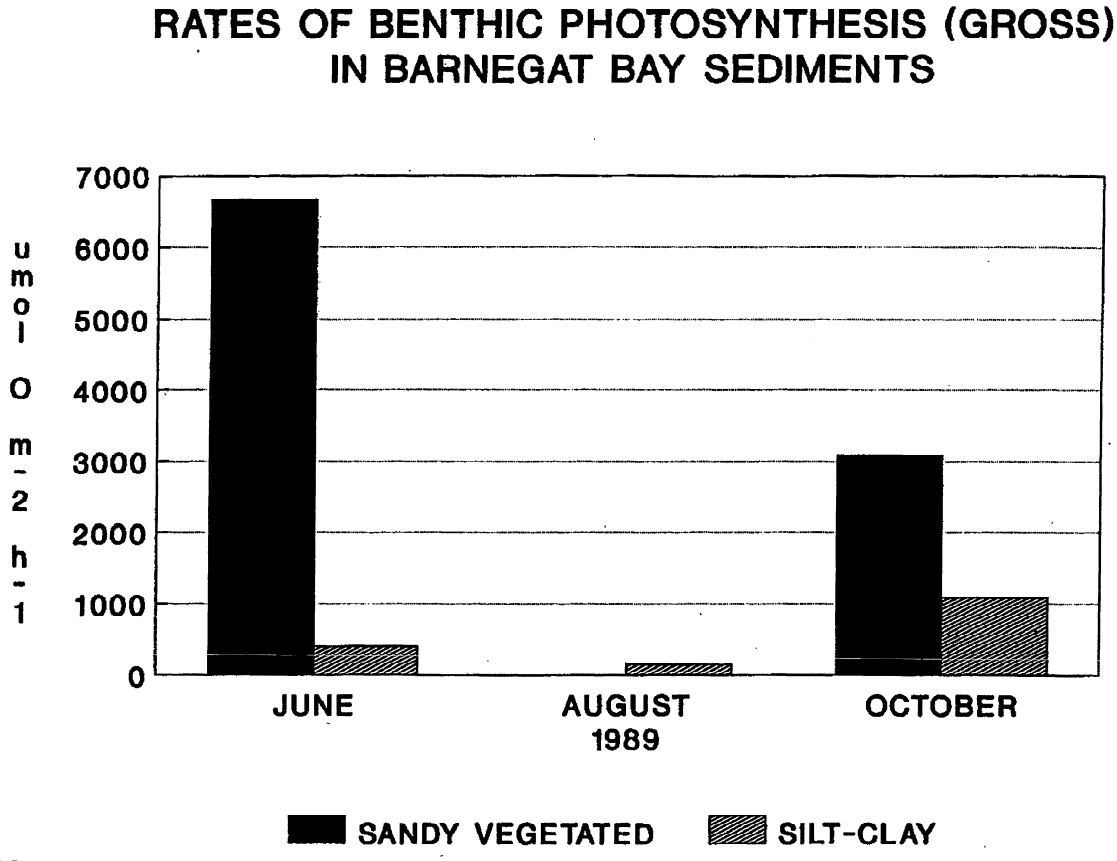


Figure 10. Comparison of the rate of benthic (gross) photosynthesis at the sandy vegetated and silt-clay site in Barnegat Bay in June, August and October 1989.

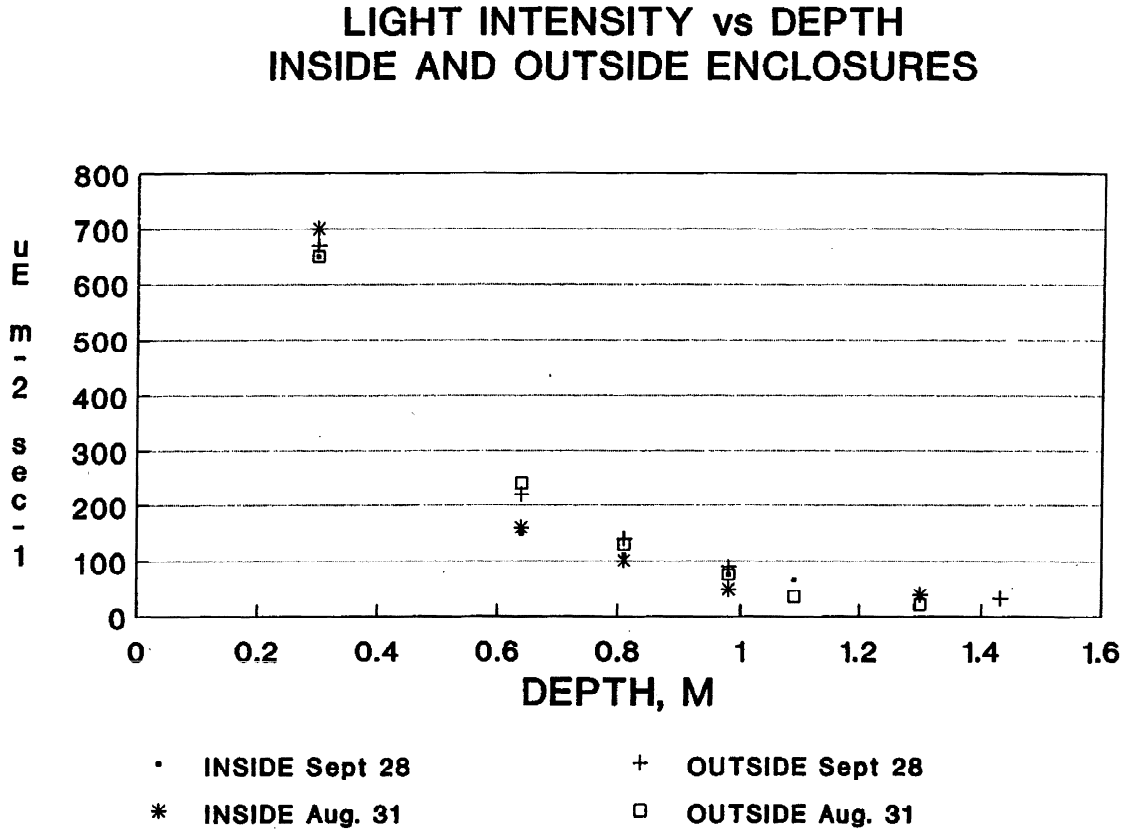


Figure 11. Comparison of light profiles (μE m^{-2} s^{-1}) inside and outside the water column enclosures used for organic N, P and C deposition measurements.

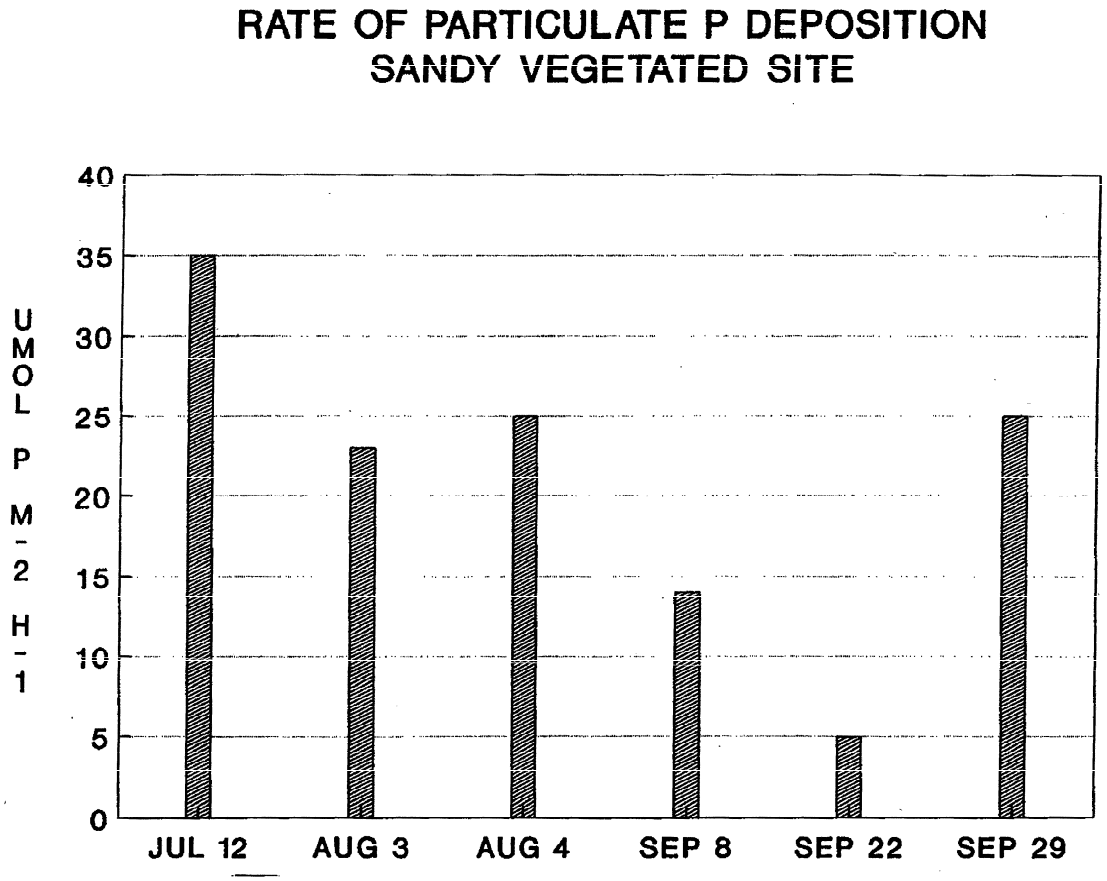


Figure 12. Rate of particulate P deposition at sandy vegetated site in Barnegat Bay measured on six days in 1989.

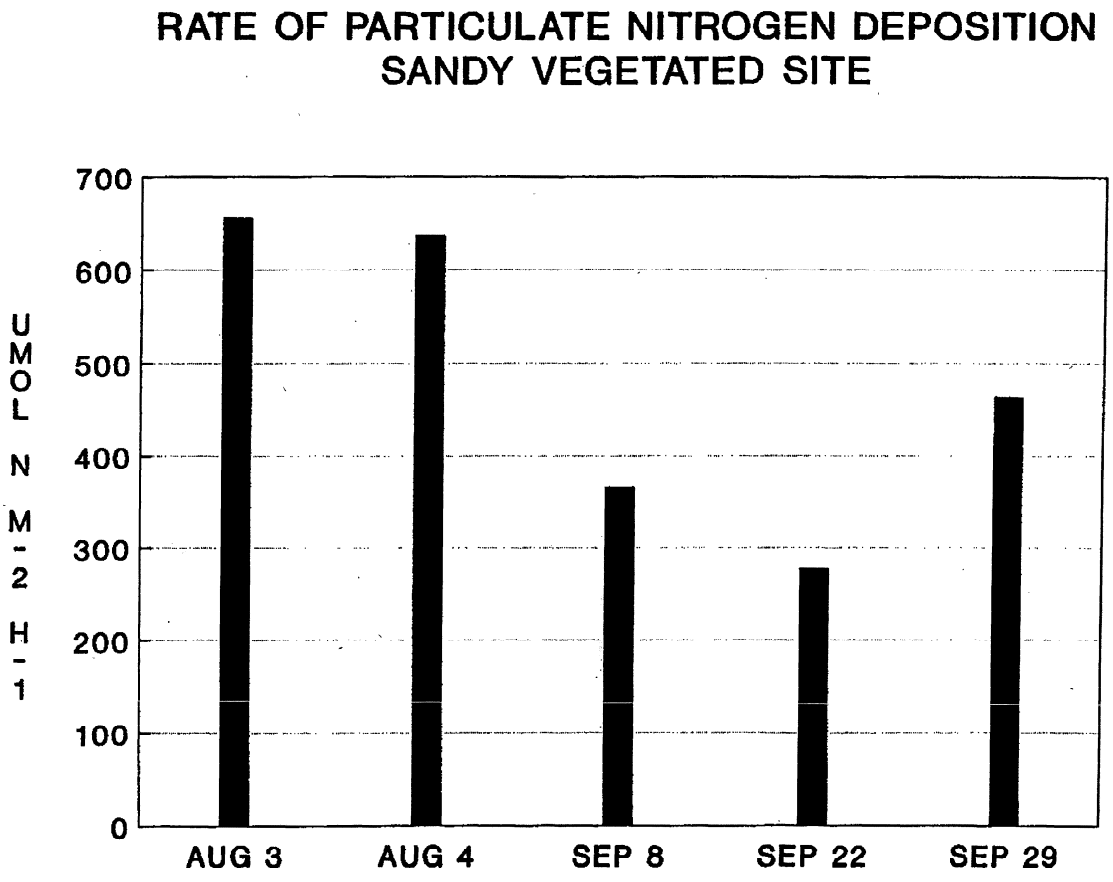


Figure 13. Rate of particulate N deposition at sandy vegetated site in Barnegat Bay measured on five days in 1989.

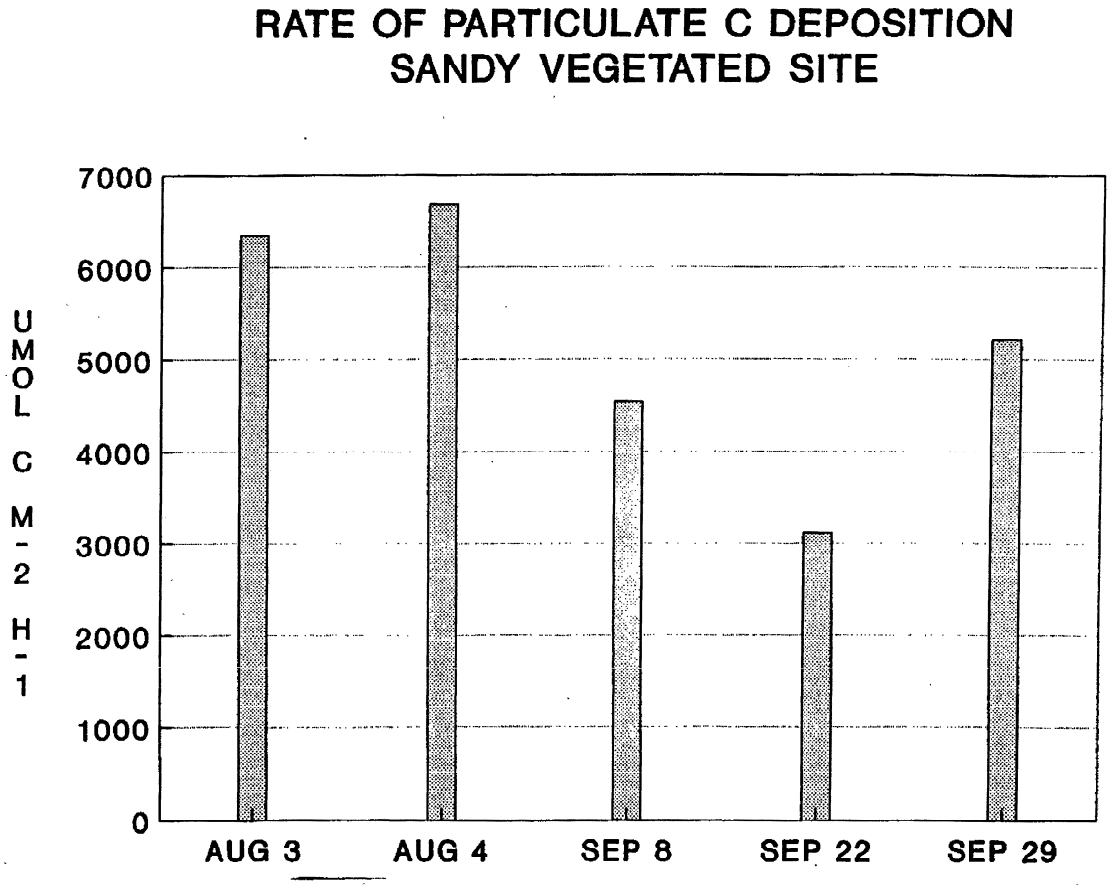


Figure 14. Rate of particulate C deposition at sandy vegetated site in Barnegat Bay measured on five days in 1989.

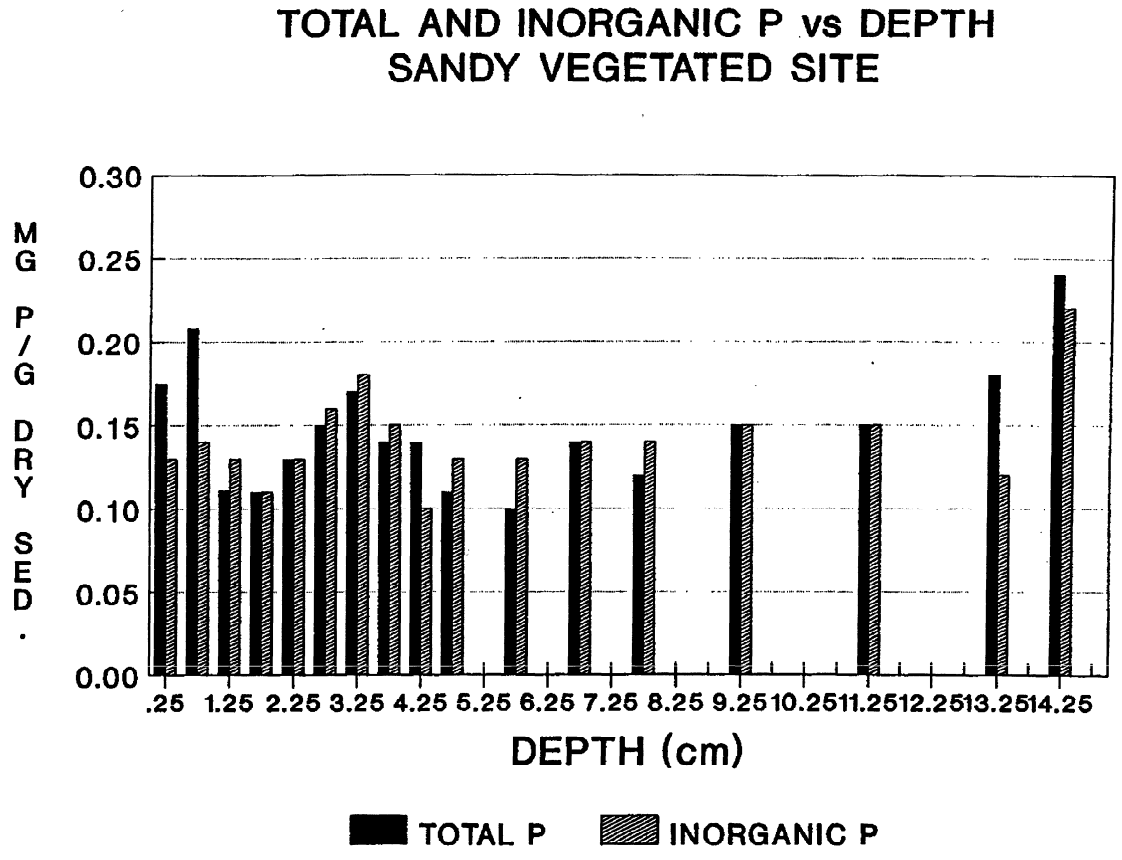


Figure 15. Total and inorganic P concentrations at various depths in sediment core collected from sandy vegetated site.

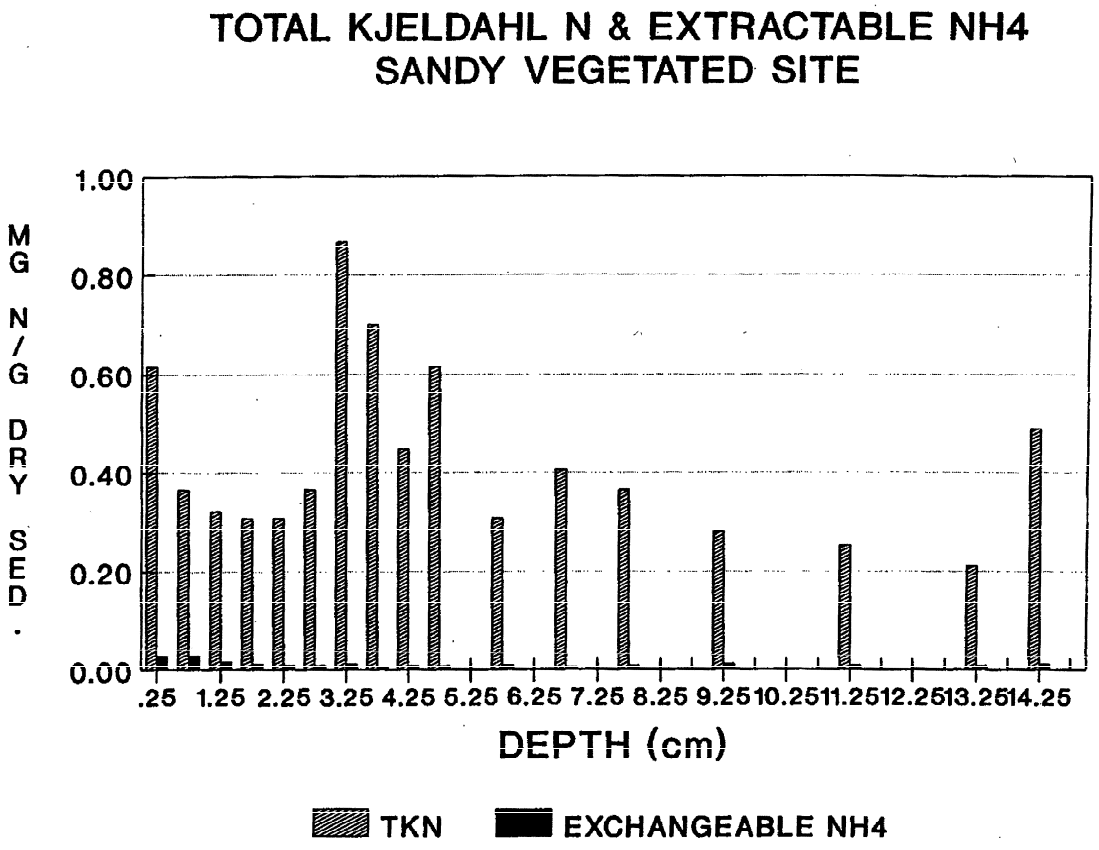


Figure 16. Total Kjeldahl N and extractable ammonia concentrations at various depths in sediment core collected from sandy vegetated site.

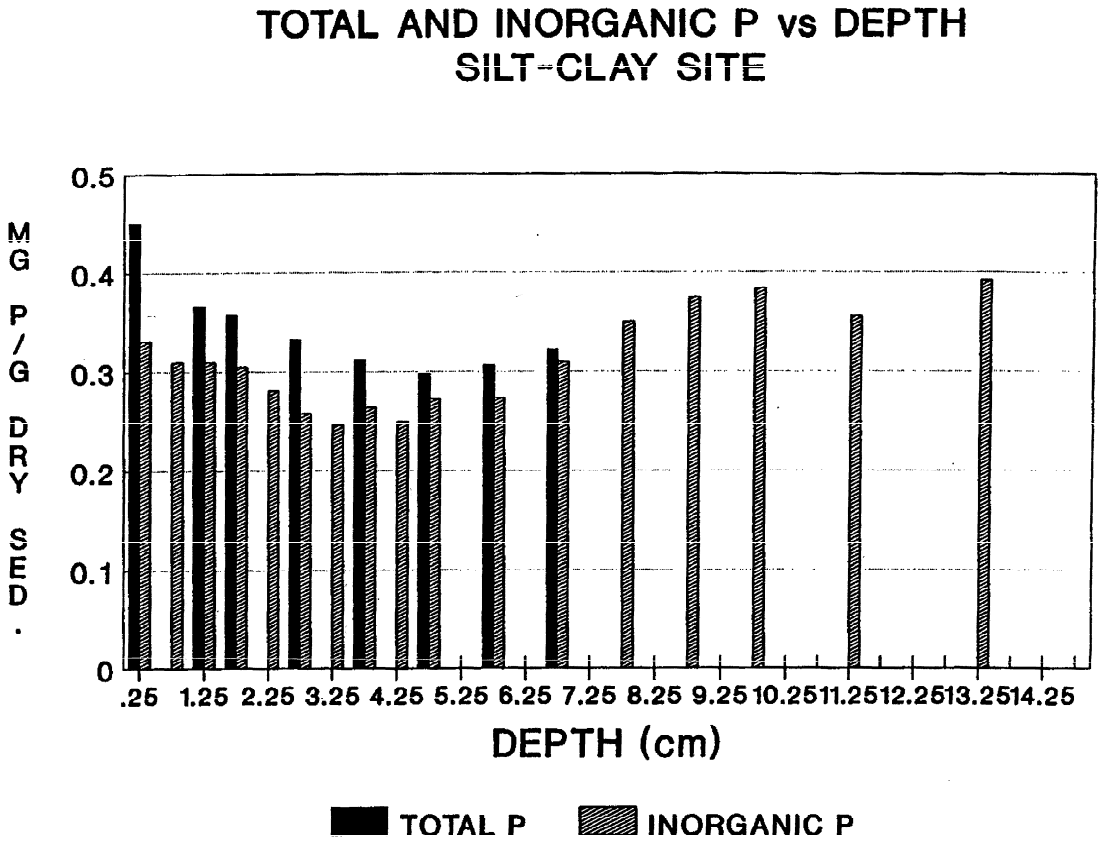


Figure 17. Total and inorganic P concentrations at various depths in sediment core collected from silt-clay site; note total P only measured at eight depths.

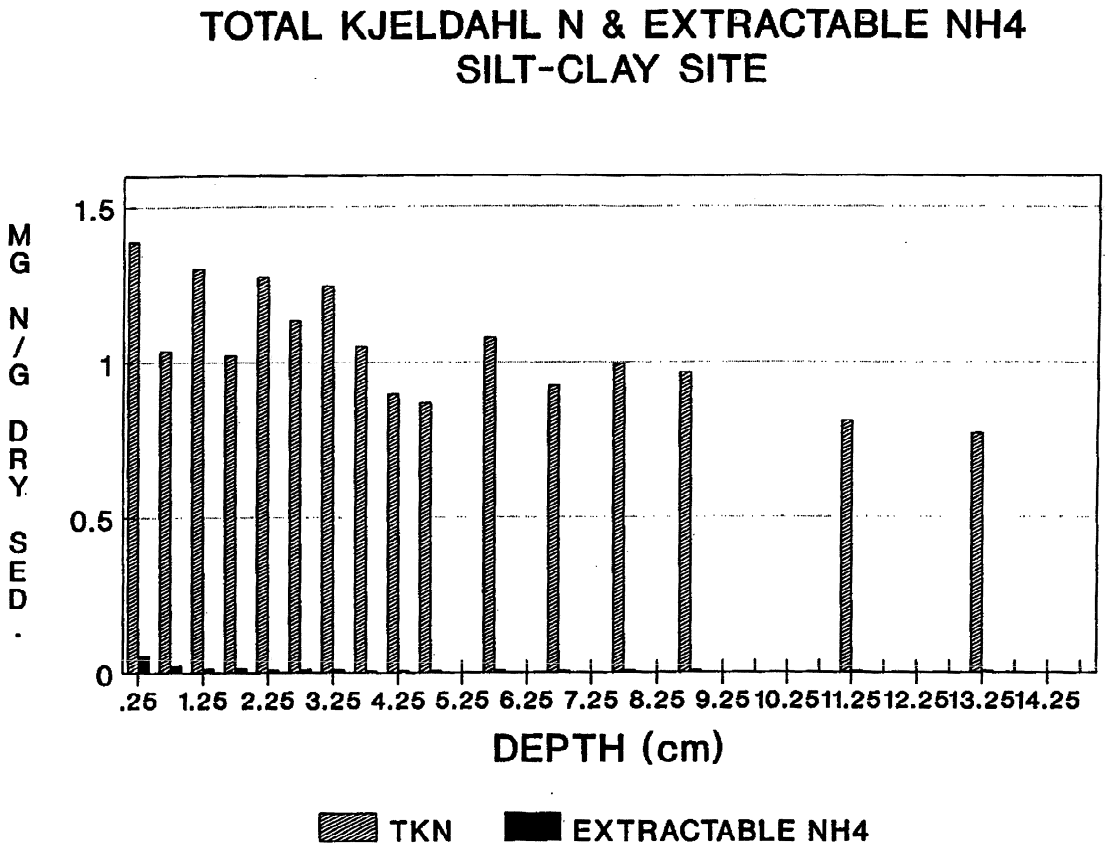


Figure 18. Total Kjeldahl N and extractable ammonia concentrations at various depths in sediment core collected from silt-clay site.

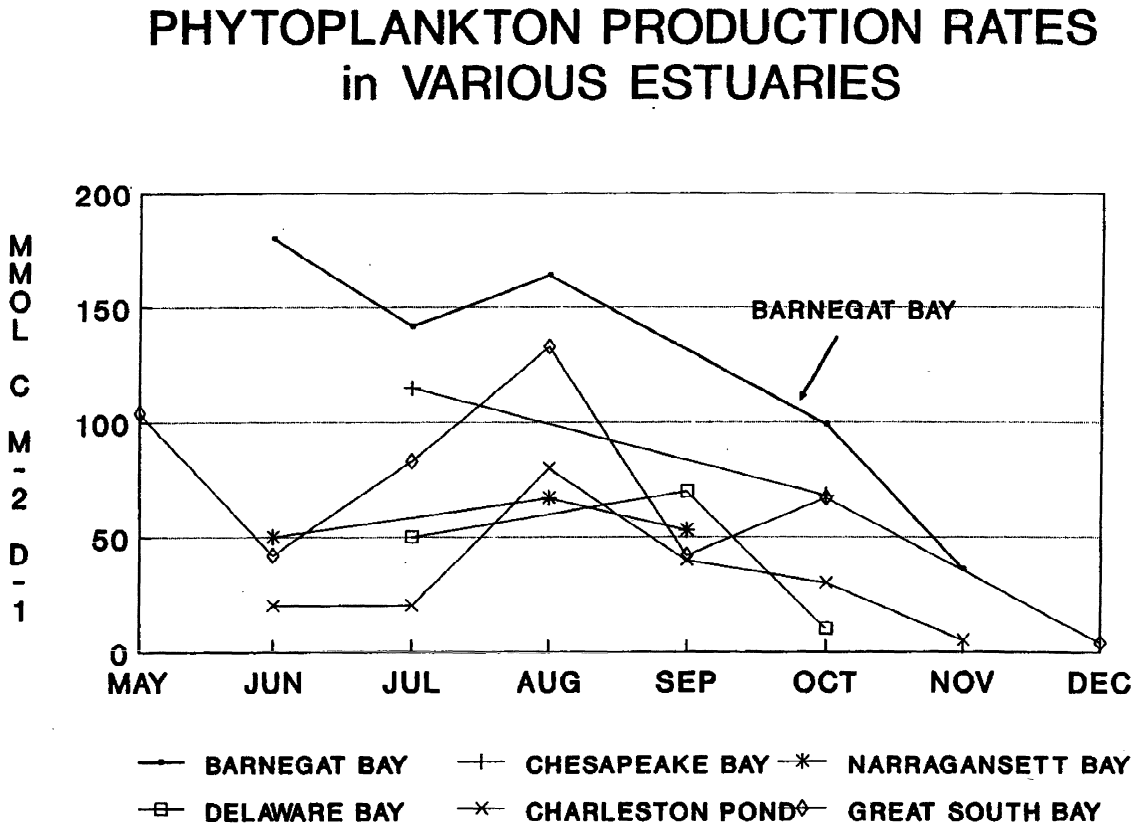


Figure 19. Phytoplankton production rates (mmol C m^{-2} d^{-1}) in various estuaries during summer. Data for Chesapeake Bay from Harding et al. (1986), Narragansett Bay from MERL (unpubl. data), Delaware Bay from Pennock and Sharp (1986), Charleston Pond, a RI coastal lagoon from Nixon and Lee (1981), and Great South Bay, LI, from Lively et al. (1986), and from Barnegat Bay in this study, calculated from NJDEPE unpubl. data as the average of four stations (see Fig. 6B).

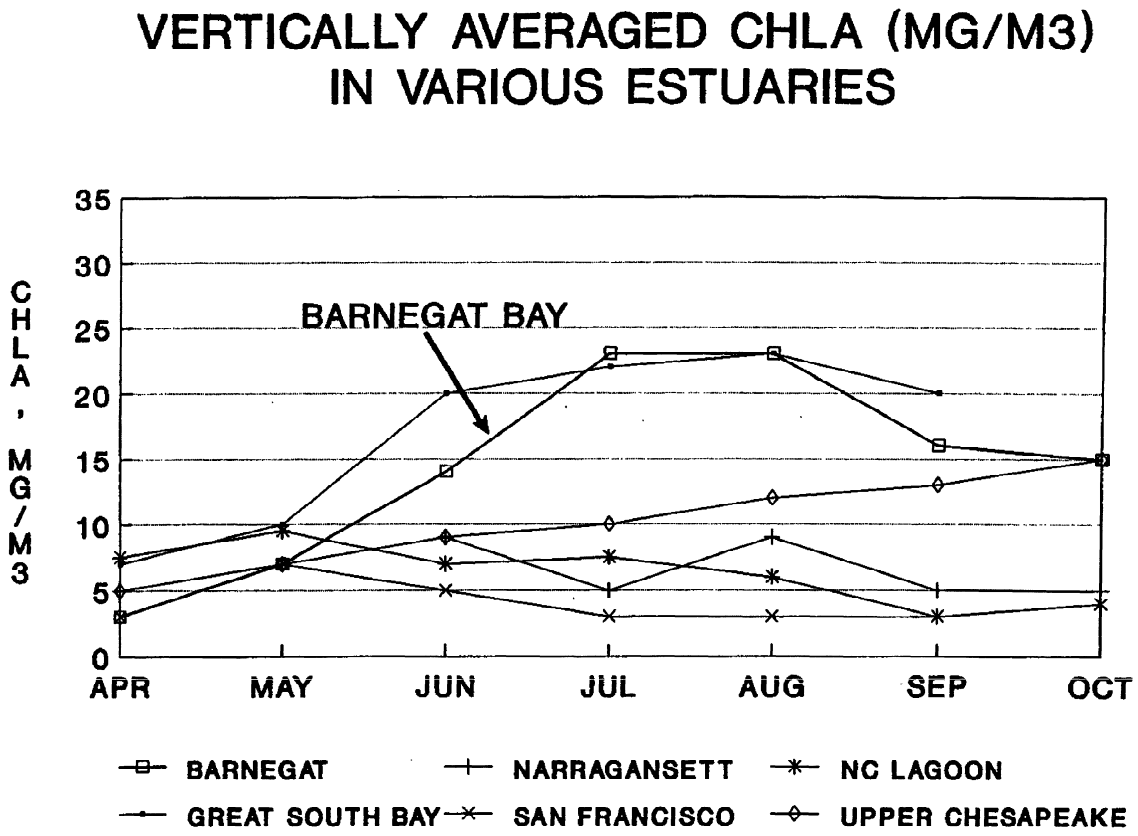


Figure 20. Comparison of vertically averaged chlorophyll *a* concentrations (mg/m^3) in various estuaries. Data for Narragansett Bay and San Francisco Bay from Nixon (1983), a North Carolina coastal lagoon from Thayer (1971), Great South Bay, LI, from Lively et al. (1986) and Upper Chesapeake Bay from Harding et al. (1986), and from Barnegat Bay in this study, calculated from NJDEPE unpubl. data as the average chlorophyll for four stations (see Fig. 4).

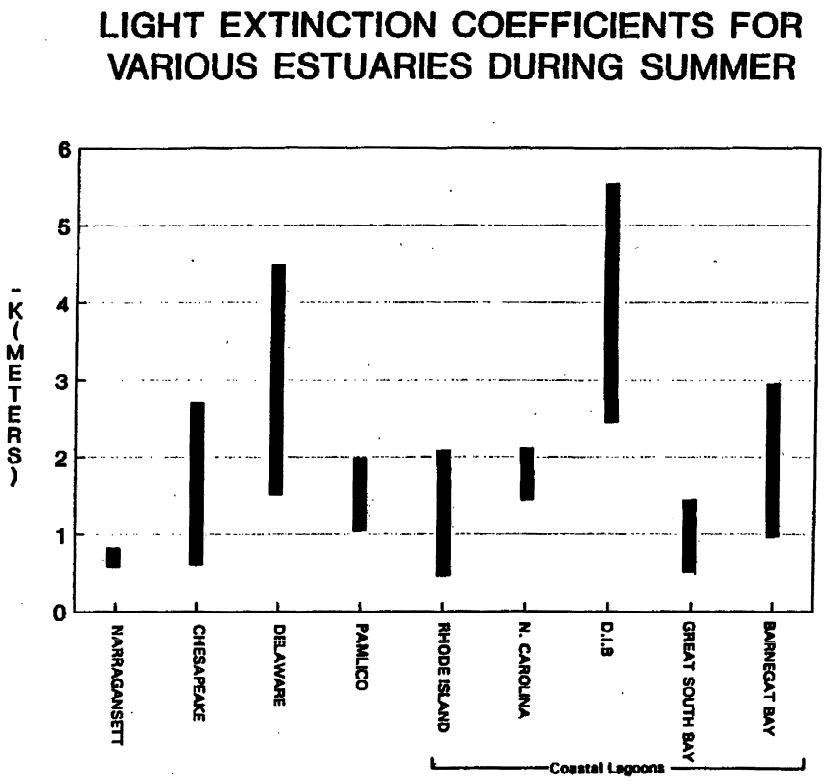


Figure 21. Light extinction coefficients ($-K_m$) in various estuaries during summer. Data for Narragansett Bay and Pamlico Sound from Nixon (1986), Chesapeake Bay from Harding et al. (1986), Delaware Bay (Culberson et al. 1987), a Rhode Island coastal lagoon, Potter Pond (annual range), from Nowicki and Nixon (1985a), open water areas of a North Carolina coastal lagoon from Thayer (1971), the Delaware Inland Bays from Sellner et al. (1988), Great South Bay, LI, from Lively et al. (1983), and from Barnegat Bay in this study, calculated from NJDEPE unpubl. data as the range for four stations (see Fig. 5).

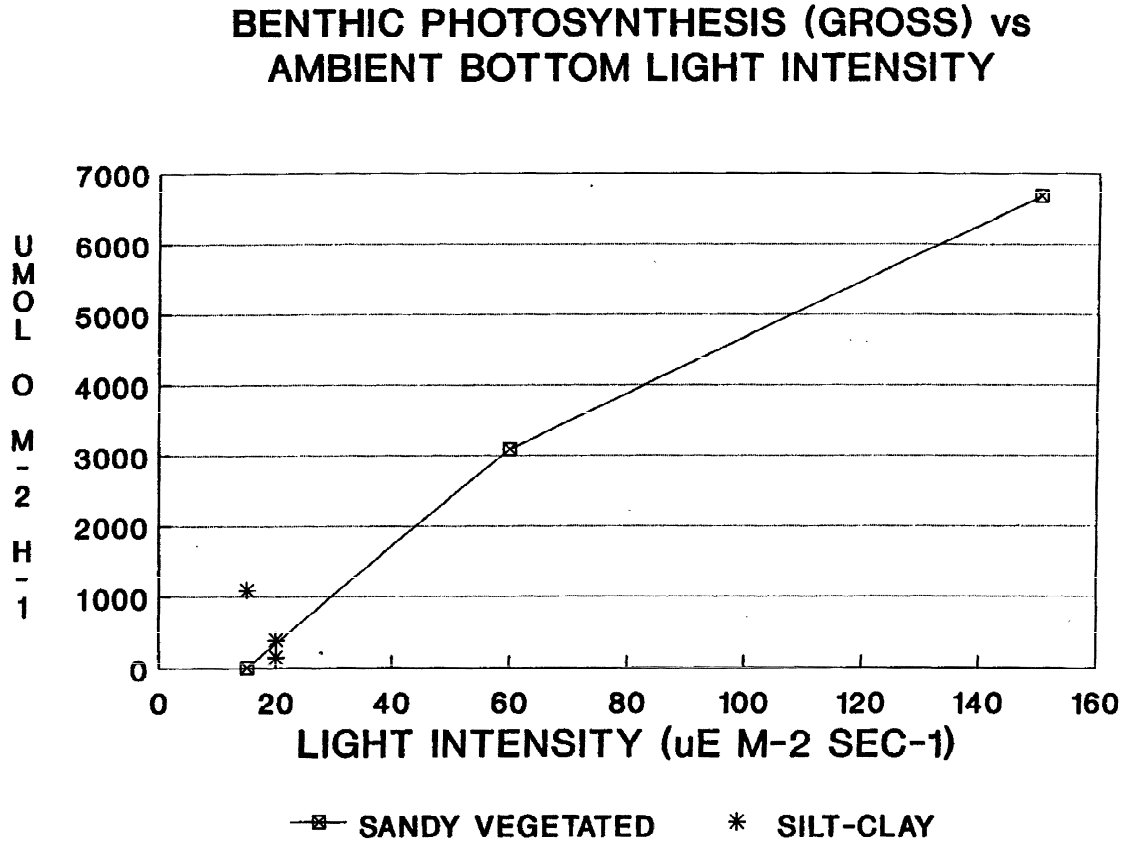


Figure 22. Rates of benthic photosynthesis (gross) as a function of ambient Bay bottom light intensity at the sandy vegetated and silt-clay sites. Data from June, August and October 1989 measurements.

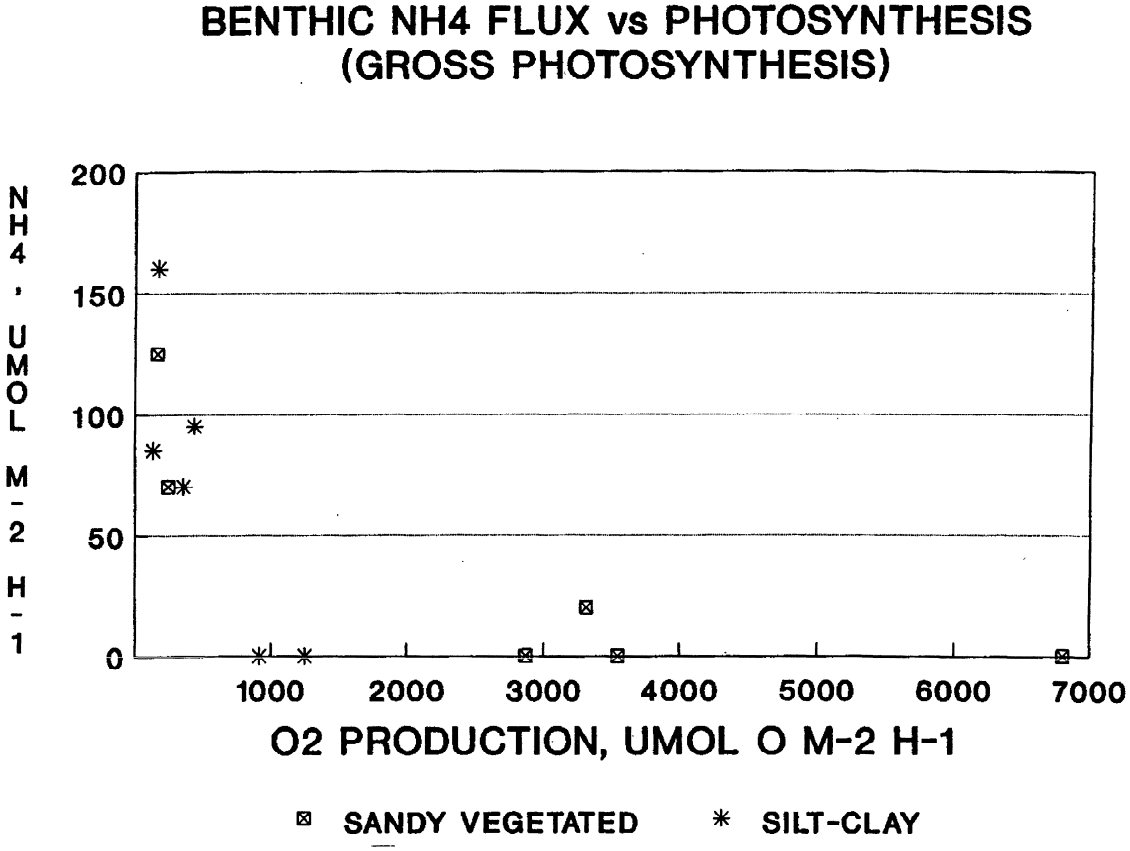


Figure 23. Sediment-water flux of ammonia vs. gross benthic photosynthesis at two locations in Barnegat Bay.

Table 1. Sediment-water fluxes (μmol $m^{-2}h^{-1}$) of oxygen, ammonia, nitrite + nitrate and phosphate measured at a sandy vegetated site and a silt-clay site in Barnegat Bay, NJ, as a function of light intensity (μE m^{-2} s^{-1}) and mid-day (gross) benthic primary production (μmol O m^{-2} h^{-1}). Predicted NH_4 and PO_4 fluxes are based on O_2 flux and Redfield stoichiometry (Eqs. 1 and 2). (n.s. = flux not significantly greater than 0 at $\rho = 0.05$; L = 100% and M = 50% of ambient bottom light intensity, and D = dark.)

Date	Core	Temp. (°C)	Salinity (‰)	Light Intensity	Measured O_2-O	Measured NH_4	Measured NO_2+NO_3	Measured PO_4	Mid-day Gross Benthic Primary Production	Predicted NH_4	Predicted PO_4
Sandy Vegetated Site											
6/21/89		25	11						6675		
	L1			150	380	n.s.	-7	n.s.		-29	-2
	L2			150	130	n.s.	n.s.	n.s.		-10	-1
	M1			75	-2350	n.s.	-9	n.s.		177	11
	M2			75	-1940	10	n.s.	n.s.		146	9
	D1			0	-6380	n.s.	n.s.	n.s.		482	30
	D2			0	-6460	n.s.	n.s.	n.s.		488	30
8/22/89		25	10						0		
	L1			15	-1810	125	n.s.	n.s.		137	9
	L2			15	-1730	70	n.s.	n.s.		131	8
	M1			7	-2300	75	n.s.	n.s.		174	11
	M2			7	-2420	140	n.s.	n.s.		183	11
	D1			0	-1850	115	n.s.	n.s.		140	9
	D2			0	-2090	150	n.s.	n.s.		158	10
10/4/89		18.5	12						3090		
	L1			60	1060	n.s.	n.s.	n.s.		-80	-5
	L2			60	1510	20	n.s.	n.s.		-114	-7
	D1			0	-1460	n.s.	n.s.	n.s.		110	7
	D2			0	-2150	n.s.	n.s.	n.s.		162	10
Silt-clay Site											
6/28/89		25	11						395		
	L1			20	-1980	95	n.s.	n.s.		149	9
	L2			20	-2060	70	n.s.	n.s.		155	10
	M1			10	-1820	85	n.s.	n.s.		137	9
	M2			10	-1350	40	n.s.	n.s.		102	6
	D1			0	-2240	175	n.s.	n.s.		169	11
	D2			0	-2590	140	n.s.	n.s.		195	12
8/15/89		25	8						150		
	L1			20	-1420	85	-5	n.s.		107	7
	L2			20	-1380	160	n.s.	n.s.		104	7
	M1			10	-1420	80	n.s.	n.s.		107	7
	M2			10	-1690	165	2	n.s.		128	8
	D1			0	-1630	150	n.s.	n.s.		123	8
	D2			0	-1470	120	n.s.	n.s.		111	7
10/4/89		18.5	12						1085		
	L1			15	-970	n.s.	n.s.	n.s.		73	5
	L2			15	-630	n.s.	n.s.	n.s.		48	3
	D1			0	-1830	n.s.	n.s.	n.s.		138	9
	D2			0	-1940	n.s.	n.s.	n.s.		146	9

Table 2. Phytoplankton samples collected from inside and outside water column enclosures at the sandy vegetated site in Barnegat Bay, 12 July 1989. Numbers are cells/ml.

	Outside Bags		Bag #1		Bag #2	
	A	B	A	B	A	B
Diatoms						
Nitzschia closterium	66	100	133	66	166	66
Coscinodiscus sp. (large >50μm)	0	0	33	0	0	33
centric diatom (small <10 μm)	2789	3851	4914	4947	4250	4482
other pennate diatoms	1527	1726	1029	797	1959	2125
Diploneis sp.	66	33	33	0	133	33
Amphora sp.	133	166	232	199	365	232
Nitzschia tryblionella	465	531	332	166	232	166
N. sigmoidea	33	0	0	33	0	100
Pleurosigma sp.	33	0	0	0	66	33
Amphipleura sp.	0	0	0	33	0	0
Thallassiosera sp. (small)	531	797	896	598	398	730
Bacillaria paradoxa	0	0	0	0	133	0
Microflagellates						
Calycomonas sp.	1959	1527	2125	1793	1228	1594
undetermined microflagellates	7702	8765	5876	5478	5113	5910
Dinoflagellates						
Gymnodinium sp.	133	133	465	199	166	66
Other						
Ebria tripartia	66	0	66	33	0	33
Dictyocha fibula	2390	1826	1926	2058	2822	2556
Schizothrix calcicola/filament	100	66	100	33	66	33
Agmenellum quadruplicatum/16 cells	0	100	0	33	33	0
Total Cells/ml	**17961**	**19621**	**18160**	**16466**	**17130**	**18192**

Table 3. Rates of mid-day (gross) phytoplankton production (mg O_2 m^{-3} h^{-1}) inside and outside water column enclosures which were used for organic matter deposition (sediment trap) studies. SV = sandy vegetated site; SC = silt-clay site.

DATE	LOCATION	OUTSIDE	INSIDE
3-Aug-89	SV	320	280
4-Aug	SV	270	270
9-Aug-89	SV	380	380
31-Aug-89	SC	350	300
1-Sep-89	SC	290	390
8-Sep-89	SV	290	230
8-Sep-89	SC	230	260
28-Sep-89	SV	270	250
29-Sep-89	SV	250	320

Table 4. Comparison of conditions inside and outside water column enclosures approximately three days after enclosure deployment.

Parameter	Outside	Inside
Nutrient Concentration		
NH_4^+, μM	<0.25	<0.25
NO_3^-, μM	0.44	0.42
$PO_4^{\equiv}$, μM	0.15	0.11
Phytoplankton		
Total Cells ml^{-1}	18,791 (±1,173)	17,488 (±840)
Phytoplankton Production		
mg C $m^{-3}h^{-1}$	24 (±7)	26 (±6)

Table 5. Rates (μmol m^{-2} h^{-1}) of deposition of particulate nitrogen, phosphorus and carbon to Barnegat Bay sediments. B1, B2 refer to replicate enclosures; two B1 or B2 on same day at same site refer to replicate sediment traps in an enclosure.

Date	Comments	N	P	C	N:P	C:N	C:P
Sandy Vegetated Site							
12-Jul-89	B1		25				
12-Jul-89	B1		40				
12-Jul-89	B2		41				
3-Aug-89	B1	656	23	6350	29	10	276
3-Aug-89	B2*						
4-Aug-89	B1	637	25	6680	25	10	267
10-Aug-89	B1*						
13-Aug-89	B1*						
8-Sep-89	B1	390	13	5090	30	13	392
8-Sep-89	B2	342	15	4015	23	12	268
14-Sep-89	B1*						
14-Sep-89	B2*						
14-Sep-89	B3*						
14-Sep-89	B4*						
22-Sep-89	B1	278	5	3115	56	11	623
22-Sep-89	B2*						
29-Sep-89	B1	333	18	3680	19	11	204
29-Sep-89	B2	498	49	5510	10	11	112
29-Sep-89	B2	558	8	6460	70	12	808
AVERAGE		462	24	5113	33	11	369
S.D.		146	14	1374	20	1	233
Silt-Clay Site							
10-Aug-89	B1*						
13-Aug-89	B1*						
1-Sep-89	B1	311	16	3745	19	12	234
8-Sep-89	B1	237	10	2930	24	12	293
	B2	361	25	3665	14	10	147
22-Sep-89	B1*						
22-Sep-89	B2*						
29-Sep-89	B1	636	36	7420	18	12	206
	B2	838	85	8475	10	10	100
	B2	528	10	5845	53	11	585
AVERAGE		485	30	5347	23	11	261
S.D.		226	29	2261	15	1	172

*Enclosure or sediment trap lost due to weather or vandalism.

Table 6. Sediment-water oxygen and nutrient fluxes (μmol m^{-2} h^{-1}) in various estuaries.

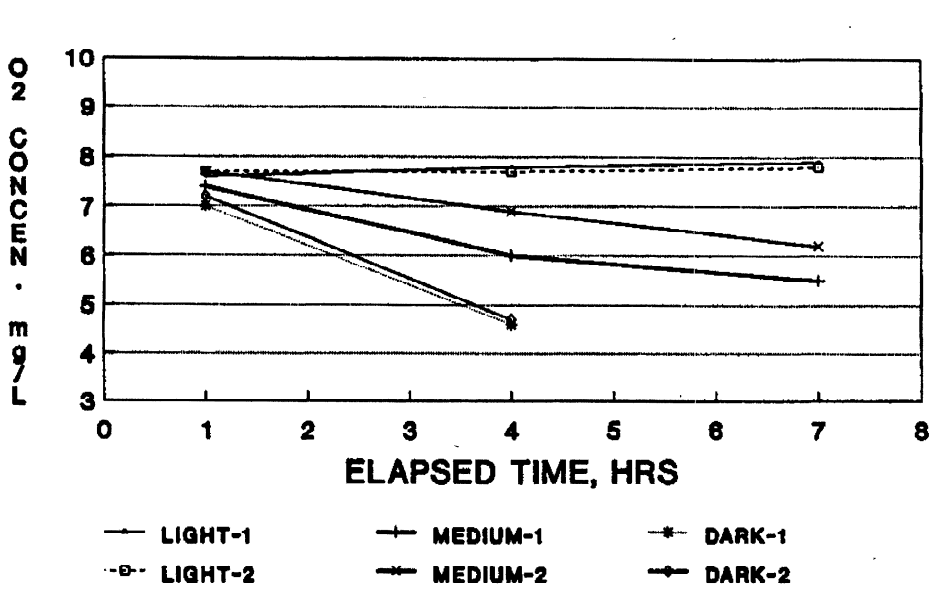
Location	Sediment Type	Date	O_2-O	NH_4 + NO_{3+2}	PO_4	Reference
Potter Pond, RI (coastal lagoon)	silt-clay and sandy	annual average	-749	42	0.6	Nowicki and Nixon 1985a,b
	silt-clay (D)	summer	-6250 to -10,950	60-440	8-50	Nowicki and Nixon 1985a,b
	sandy (D)	summer	-1550 to -4700	0-50*	0-10	Nowicki and Nixon 1985a,b
Absecon Bay, NJ	silt-clay	Sept-Oct		515*		Durand 1984
				334*		Durand 1984
South River Estuary, NC	silt-clay	annual	-3540	114	6.25	Fisher et al. 1982
Neuse River Estuary, NC	silt-clay	annual	-3020	227	14.0	Fisher et al. 1982
Ochlockonee Bay, FL	silt-clay	summer	-1083	82	< 1	Seitzinger 1987
Narragansett Bay, RI	silt-clay	summer	-4700	200	30-50	Nixon et al. 1976, 1980
Chesapeake Bay						
Upper Bay	—	August	-4166	138*	-4-2	Boynton and Kemp 1985
Mid Bay	—		-8073	563*	15-40	Boynton and Kemp 1985
Lower Bay	—		-3906	263*	10	Boynton and Kemp 1985
Georgia Bight, USA	sandy	summer	-7552	175	37	Hopkinson and Wetzel 1982
La Jolla Bight, CA	sandy	summer		40	6	Hartwig 1976
Four League Bay, LA	silt-clay	annual	-3063	110	-8	Teague et al. 1988
Bowling Green Bay Reef	silt-clay	August	—	330	1	Ullman and Sandstrom 1987
Lagoon, Australia	sandy		—	-29	-17	Ullman and Sandstrom 1987
Barnegat Bay, NJ	sandy (L)	June	205	< 10	n.s.	this study
	sandy (D)	June	-6420	n.s.	n.s.	this study
	silt-clay (D)	June	-2420	160	n.s.	this study
	sandy(D)	August	-1970	130	n.s.	this study
	silt-clay (D)	August	-1550	135	n.s.	this study

*NH_4 only

**L = at ambient mid-day bottom light intensity; D = dark

n.s. = not significantly greater than 0

A



B

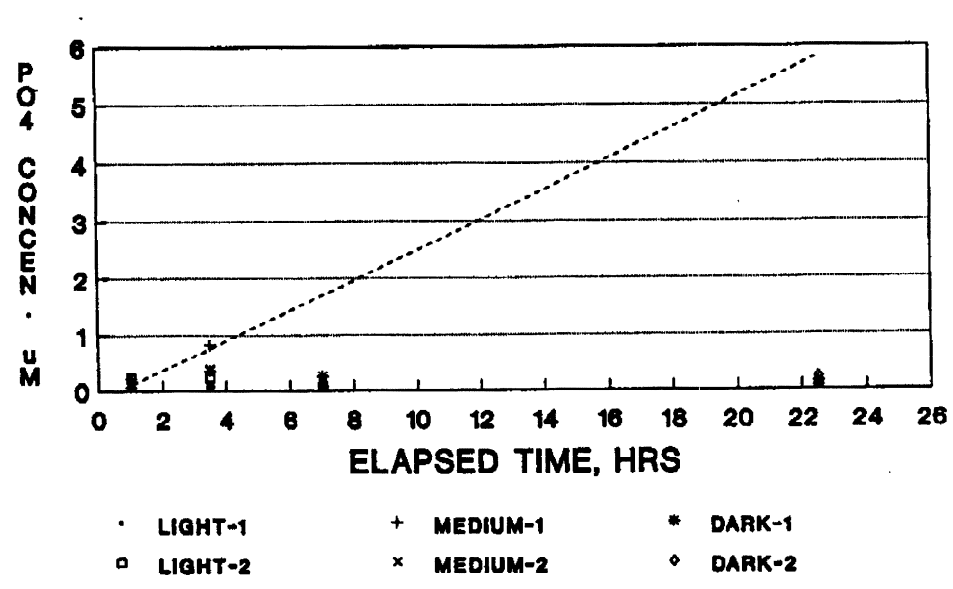
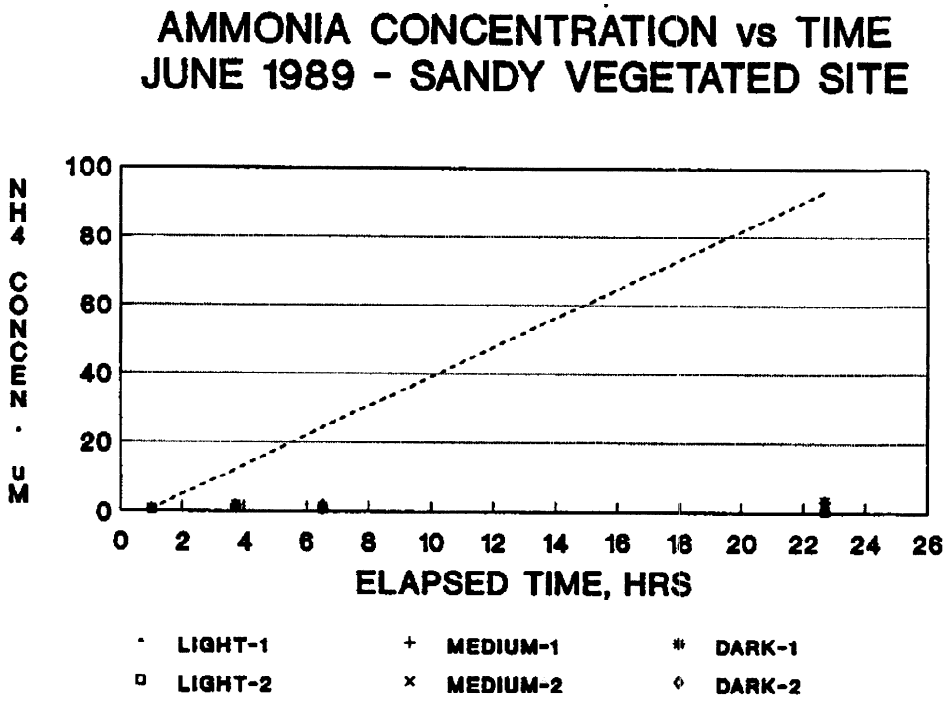


Figure A-1. A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the sandy vegetated site in Barnegat Bay, June 1989. Cores were incubated at ambient Bay bottom light intensity (light), 50% of bottom light intensity (medium), or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

C



D

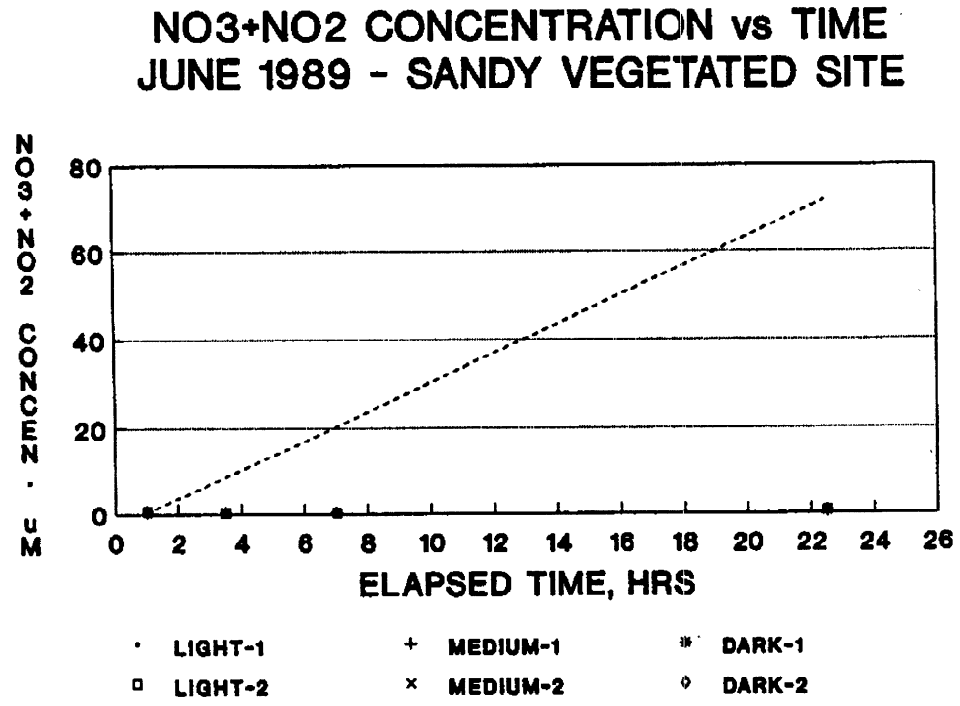
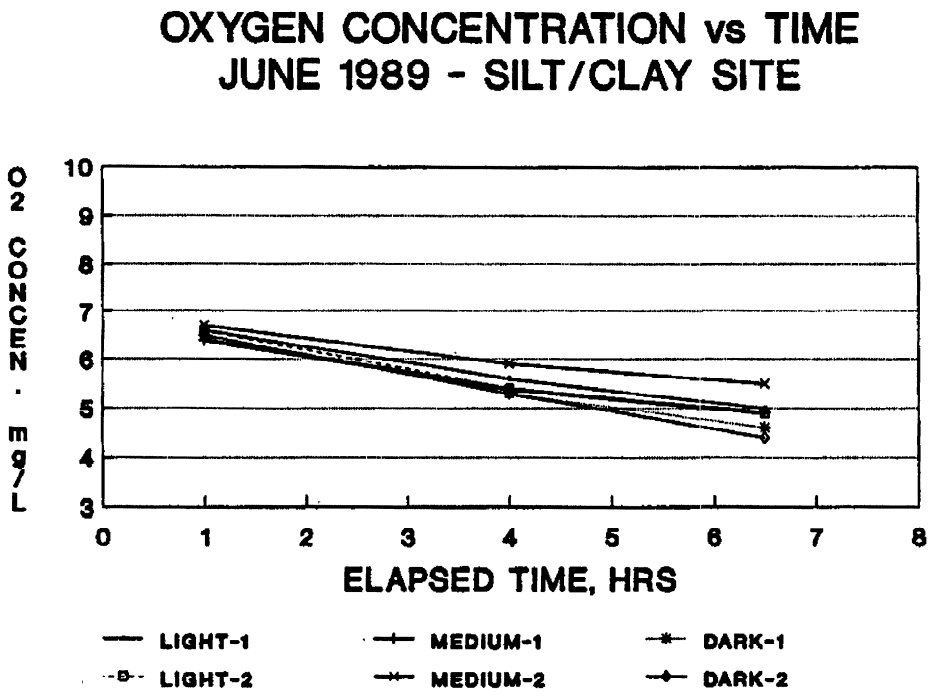


Figure A-1 (continued). A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the sandy vegetated site in Barnegat Bay, June 1989. Cores were incubated at ambient Bay bottom light intensity (light), 50% of bottom light intensity (medium), or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

A



B

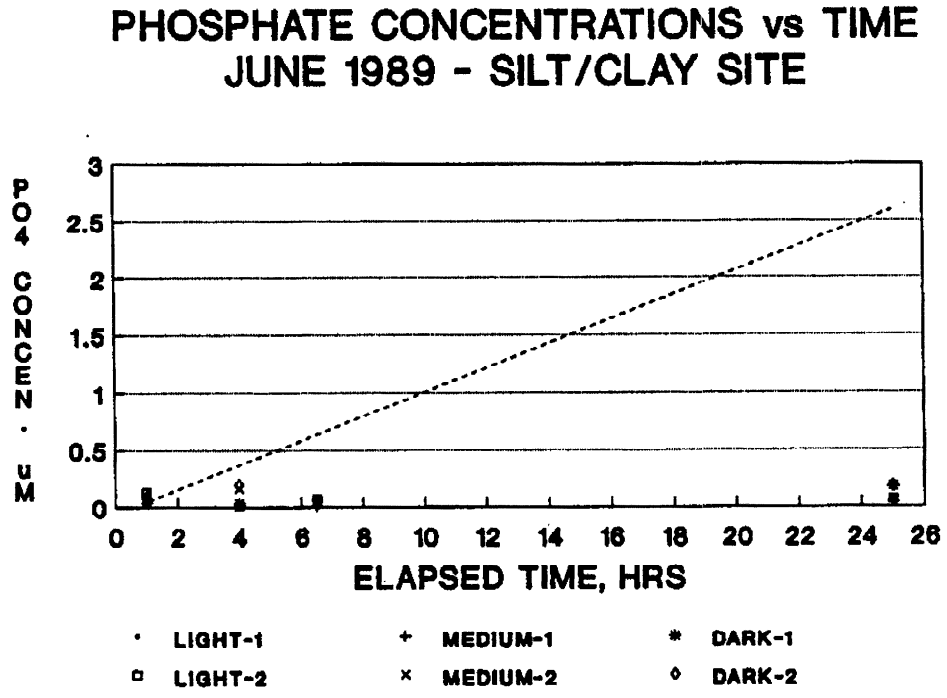
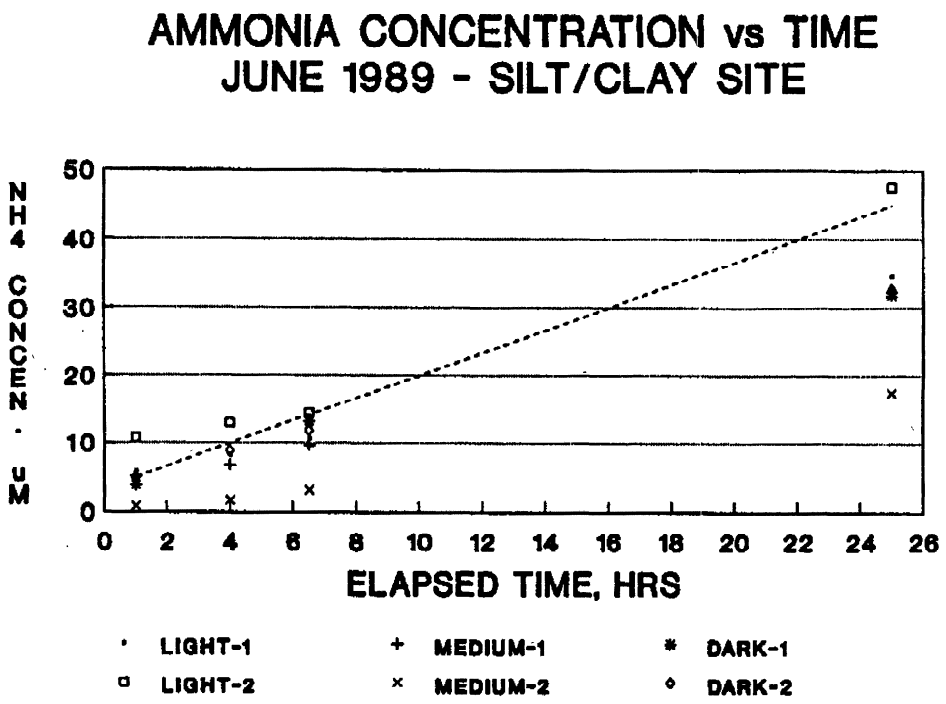


Figure A-2. A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the silt/clay site in Barnegat Bay, June 1989. Cores were incubated at ambient Bay bottom light intensity (light), 50% of bottom light intensity (medium), or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

C



D

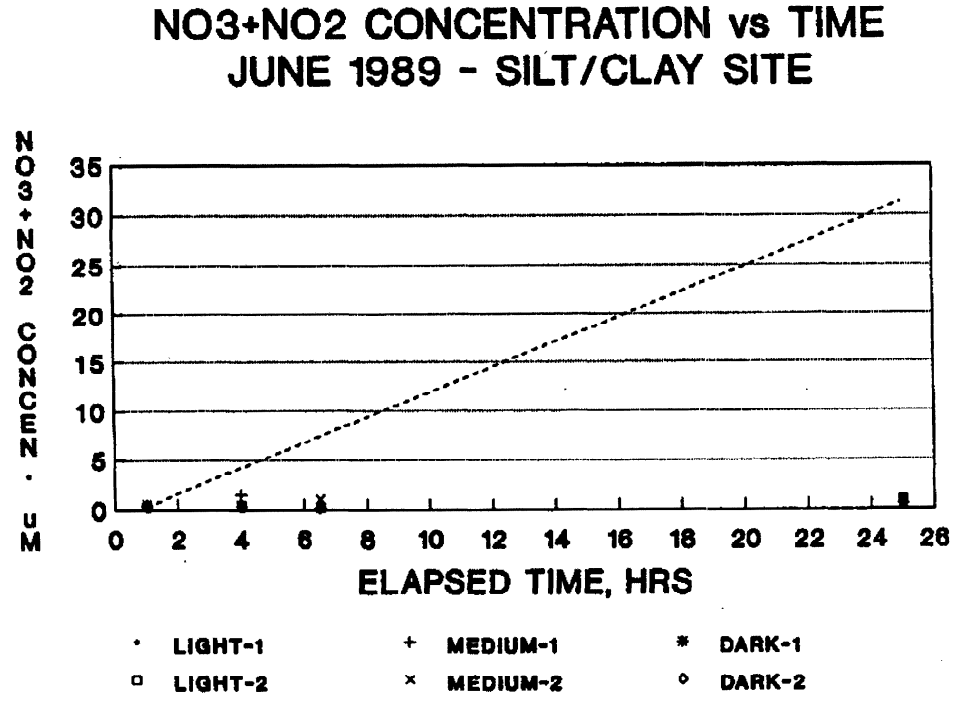
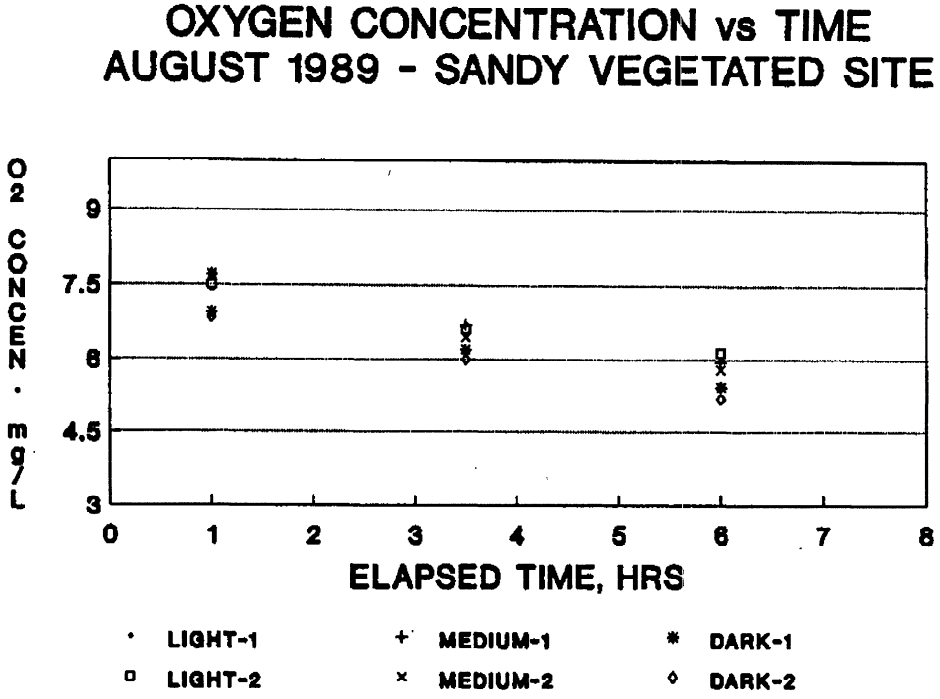


Figure A-2 (continued). A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the silt/clay site in Barnegat Bay, June 1989. Cores were incubated at ambient Bay bottom light intensity (light), 50% of bottom light intensity (medium), or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

A



B

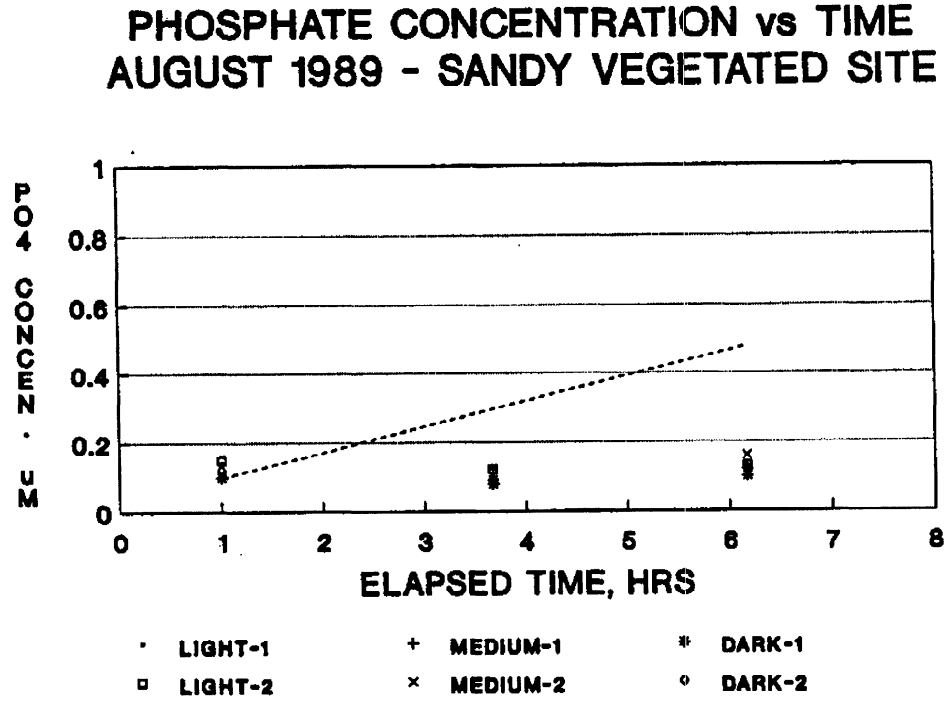
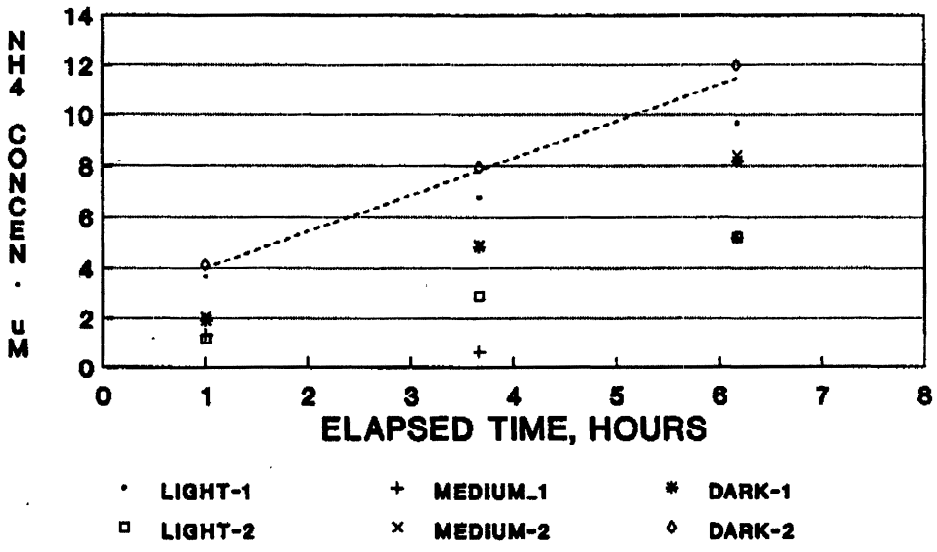


Figure A-3. A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the sandy vegetated site in Barnegat Bay, August 1989. Cores were incubated at ambient Bay bottom light intensity (light), 50% of bottom light intensity (medium), or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

C



D

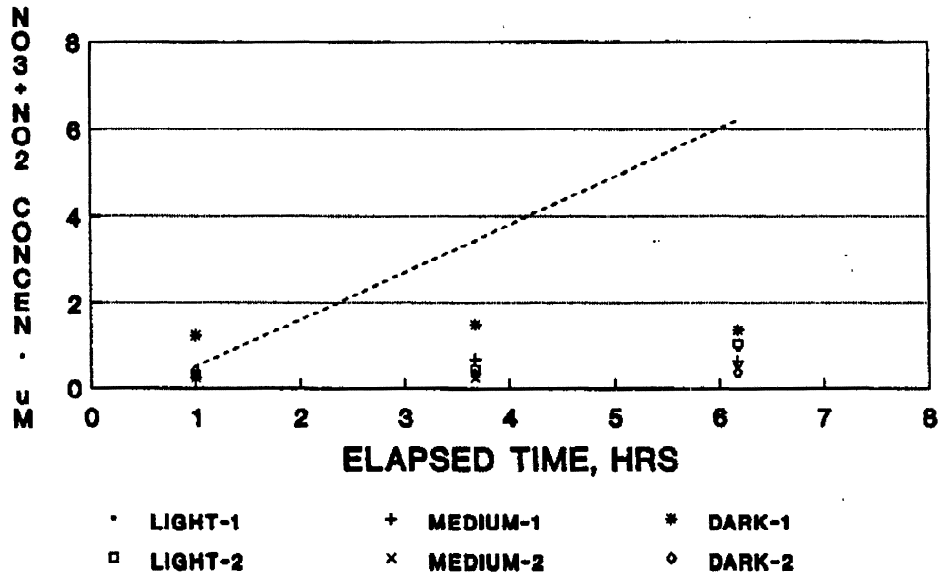
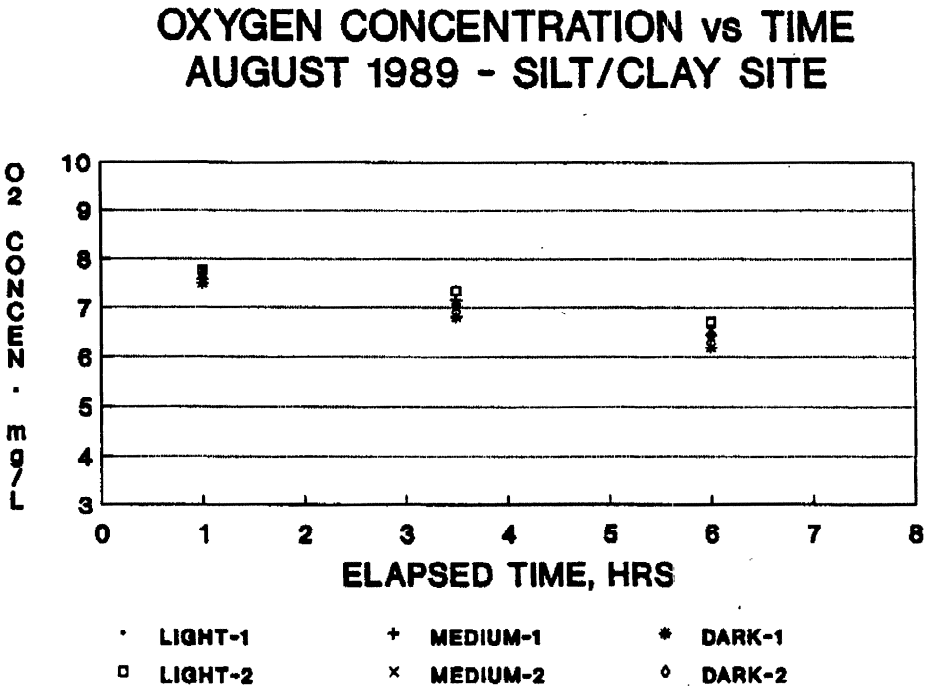


Figure A-3 (continued). A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the sandy vegetated site in Barnegat Bay, August 1989. Cores were incubated at ambient Bay bottom light intensity (light), 50% of bottom light intensity (medium), or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

A



B

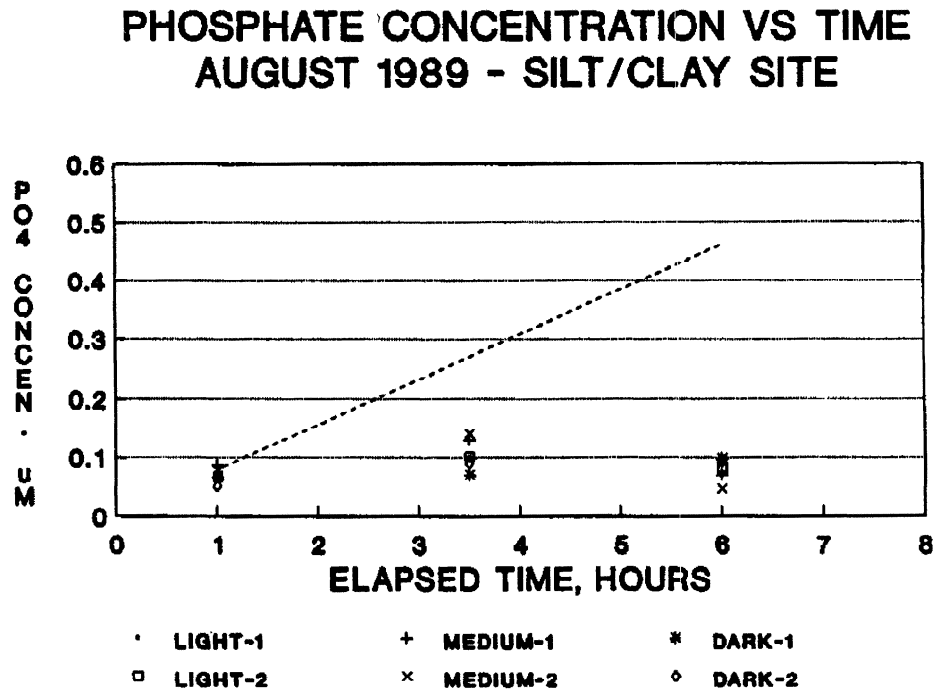
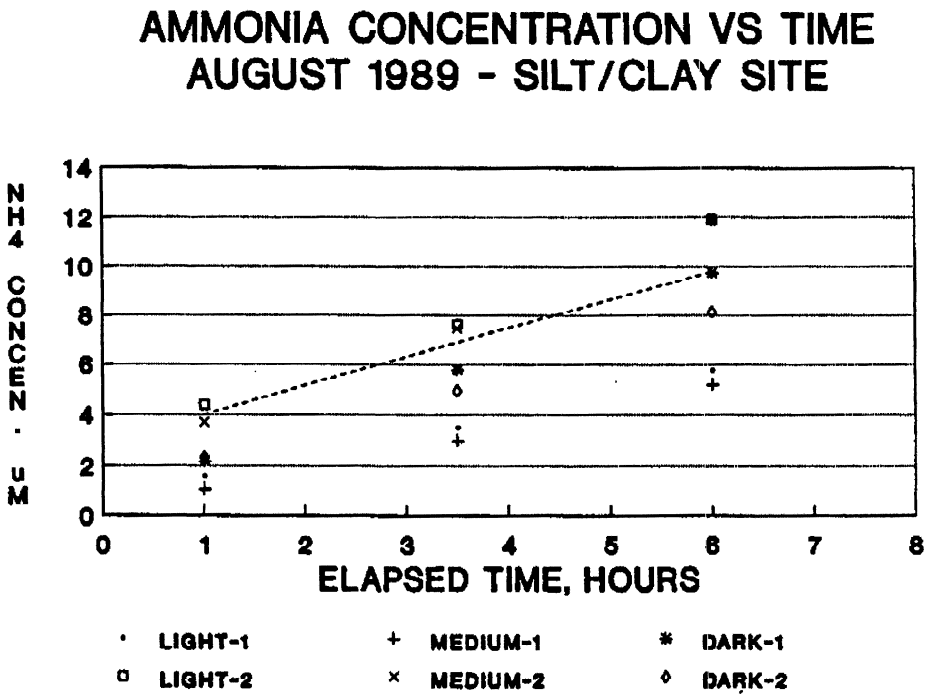


Figure A-4. A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the silt/clay site in Barnegat Bay, August 1989. Cores were incubated at ambient Bay bottom light intensity (light), 50% of bottom light intensity (medium), or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

C



D

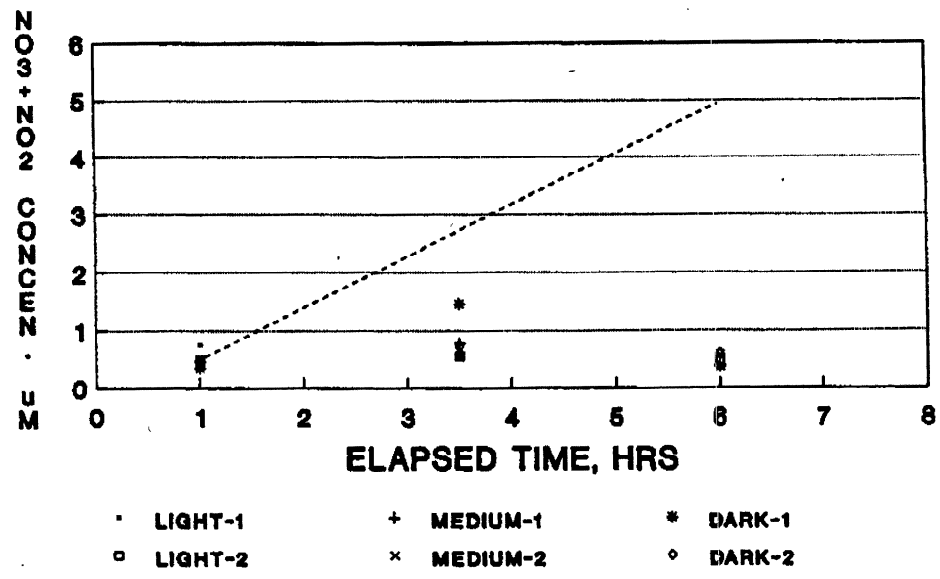
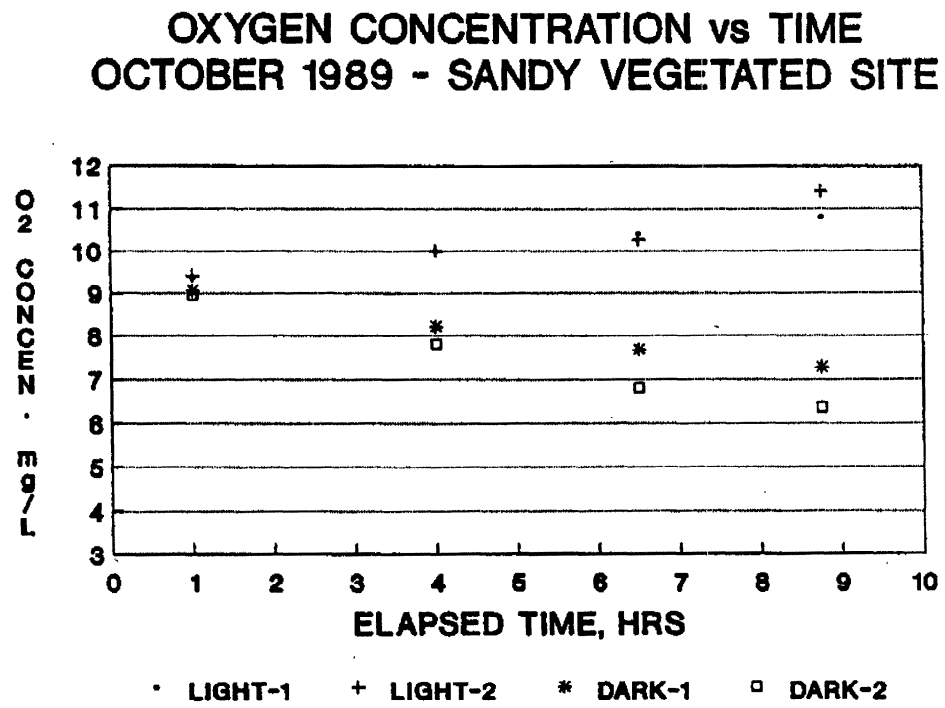


Figure A-4 (continued). A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the silt/clay site in Barnegat Bay, August 1989. Cores were incubated at ambient Bay bottom light intensity (light), 50% of bottom light intensity (medium), or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

A



B

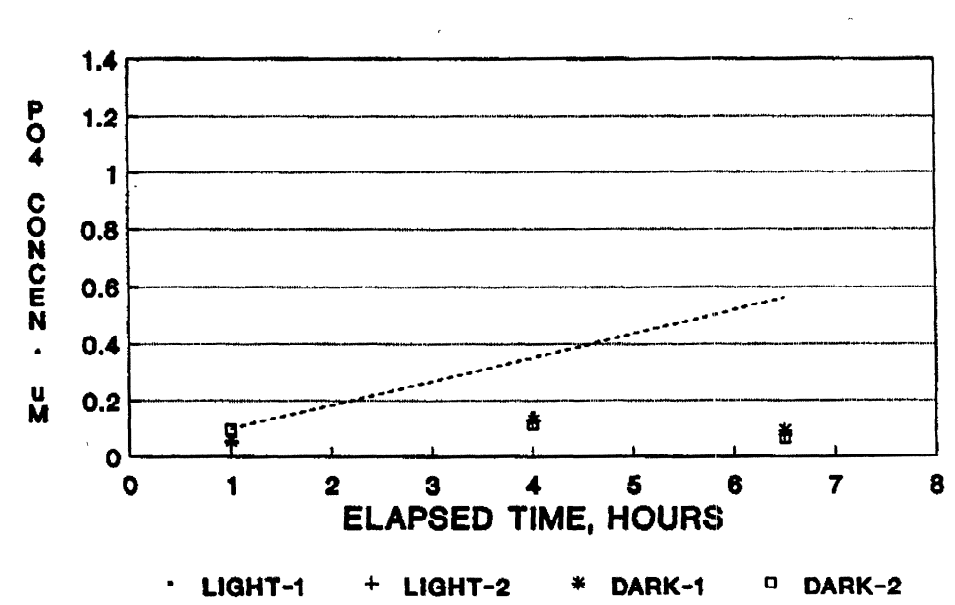
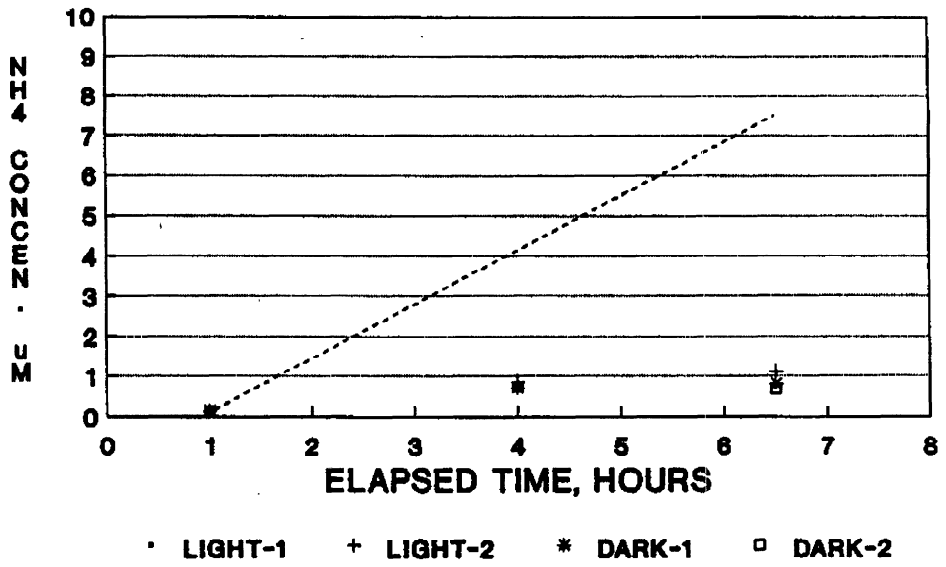


Figure A-5. A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the sandy vegetated site in Barnegat Bay, October 1989. Cores were incubated at ambient Bay bottom light intensity (light) or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

C



D

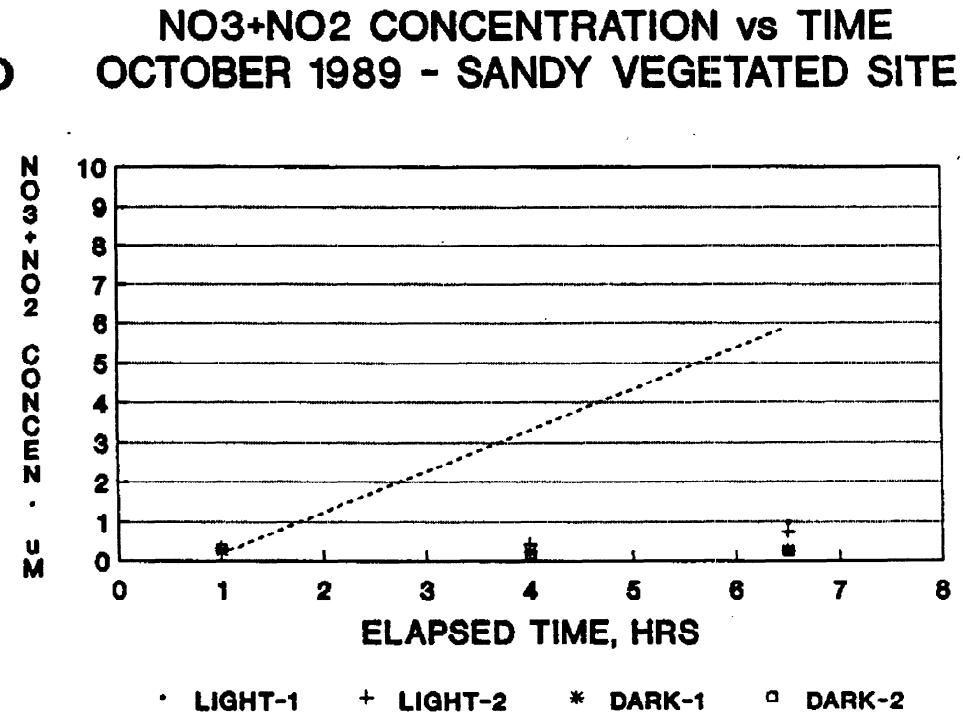
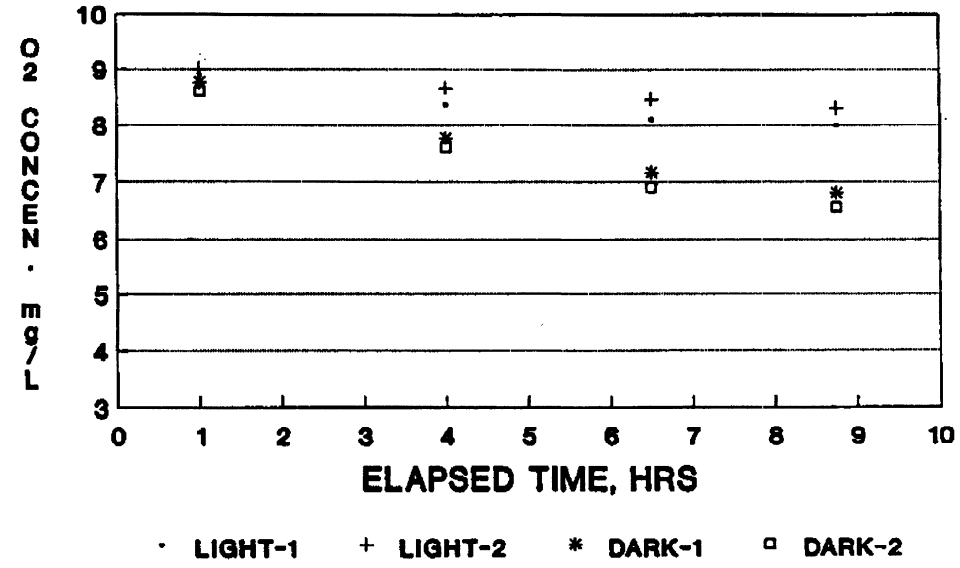


Figure A-5 (continued). A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the sandy vegetated site in Barnegat Bay, October 1989. Cores were incubated at ambient Bay bottom light intensity (light) or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

A



B

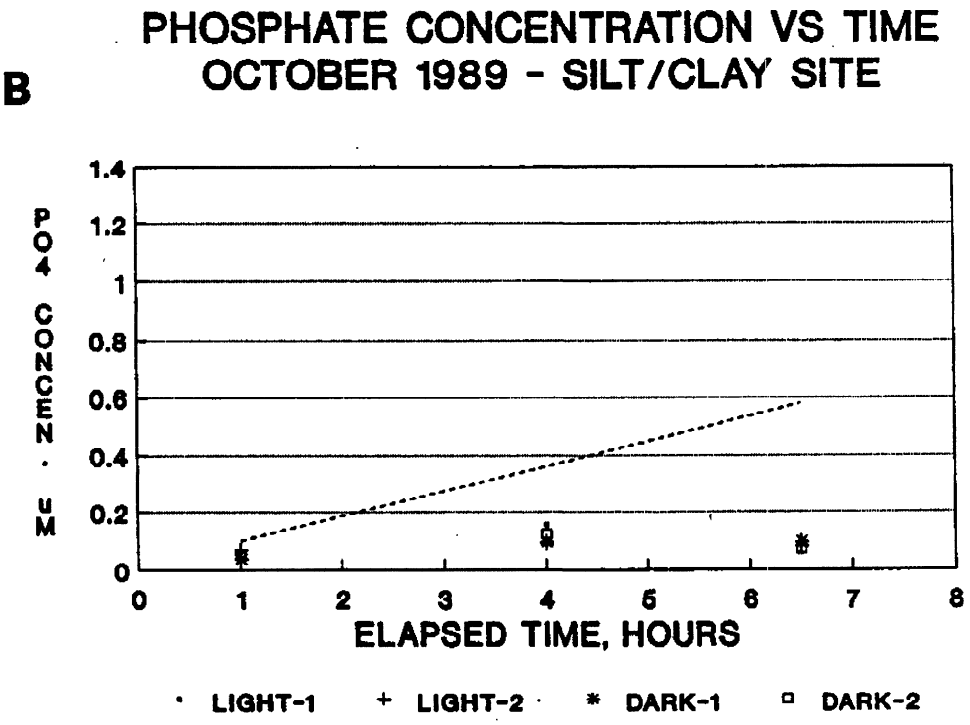
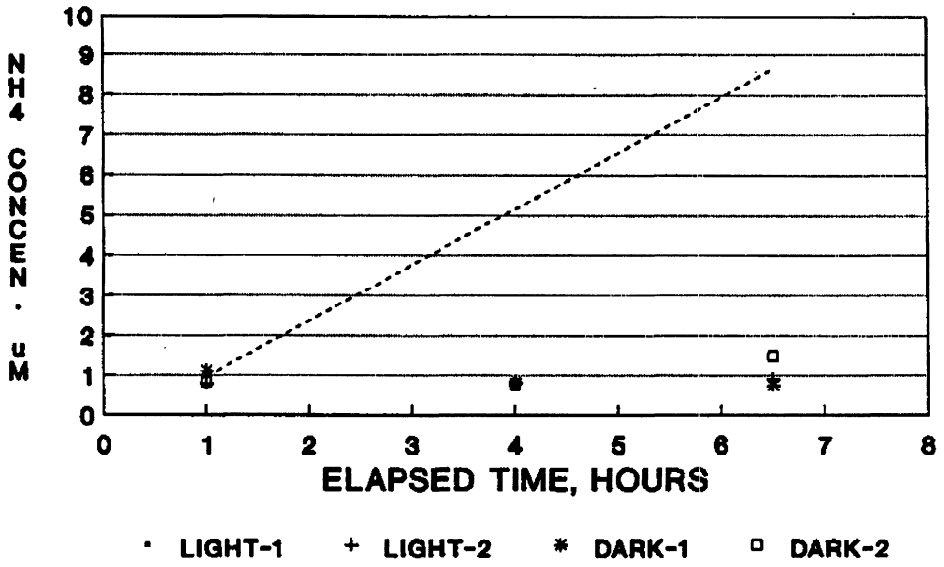


Figure A-6. A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the silt/clay site in Barnegat Bay, October 1989. Cores were incubated at ambient Bay bottom light intensity (light), or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

C



D

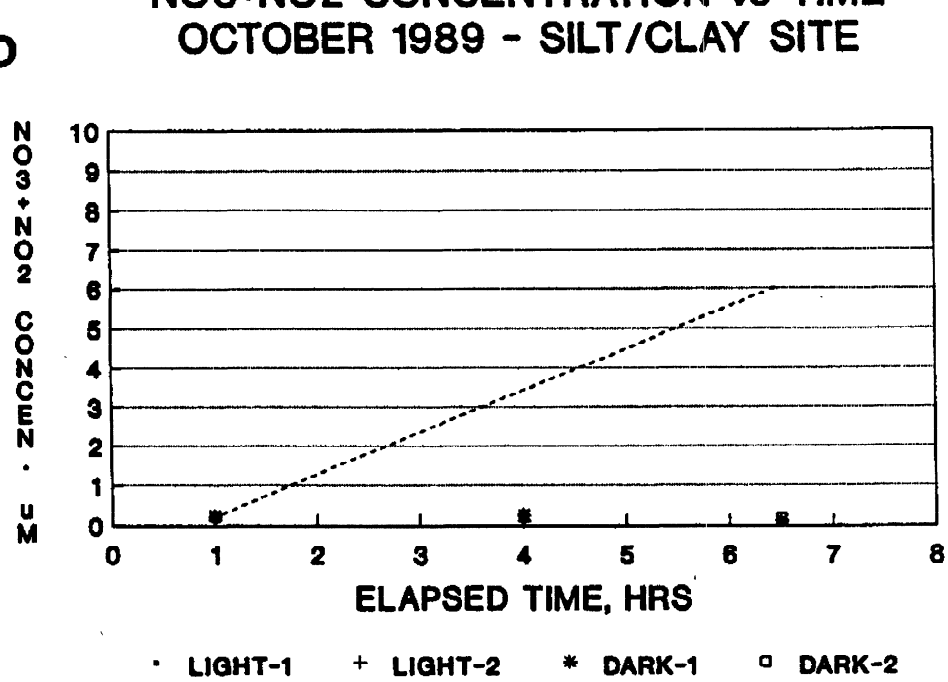
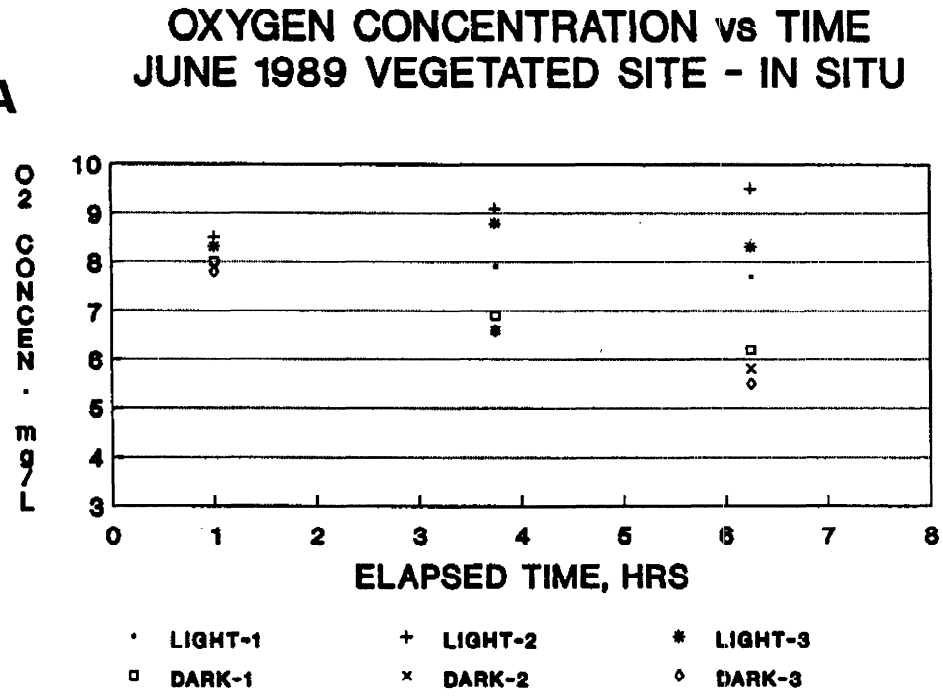


Figure A-6 (continued). A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying laboratory-incubated sediment cores from the silt/clay site in Barnegat Bay, October 1989. Cores were incubated at ambient Bay bottom light intensity (light), or in the dark (dark). The dashed line is the predicted phosphate, ammonia or nitrate plus nitrite concentration in the overlying water of dark incubated cores based on the average dark oxygen uptake rate and an O:P ratio of 212:1, O:NH_4 ratio of 13.25:1, or an O:NO_3 ratio of 17.25:1 (see text).

A



B

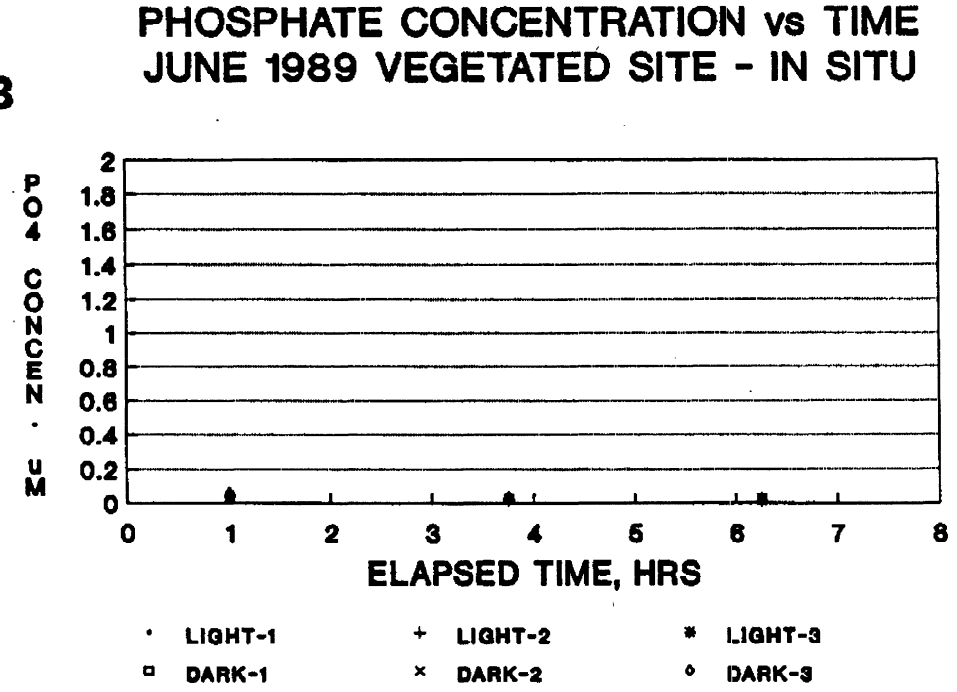
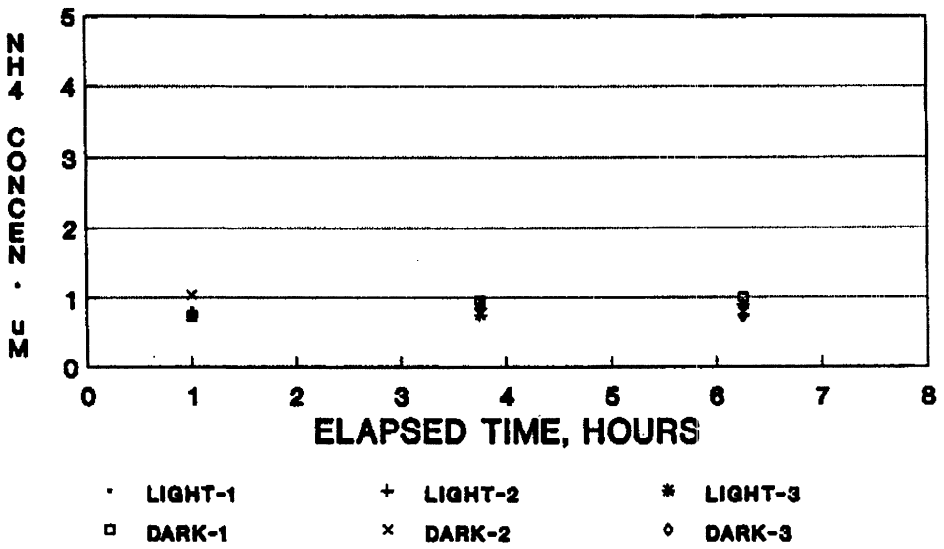


Figure A-7. A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying sediments in *in situ* benthic chambers at the sandy vegetated site in Barnegat Bay, June 1989. Cores were incubated at ambient Bay bottom light intensity (light), or in the dark (dark).

C



D

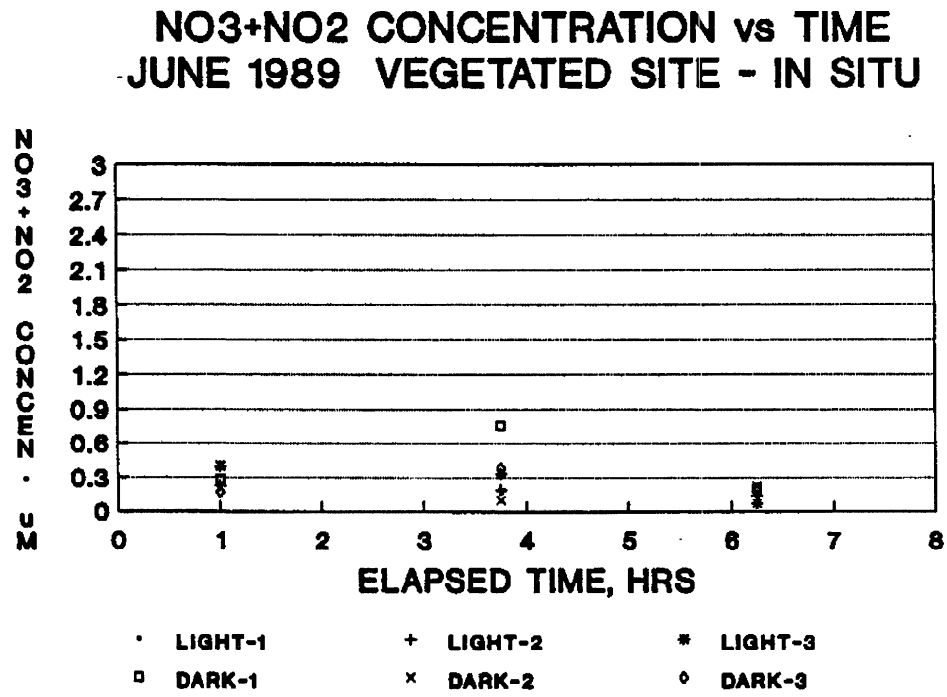


Figure A-7 (continued). A) Oxygen, B) phosphate, C) ammonia and D) nitrate plus nitrite concentration versus time in water overlying sediments in *in situ* benthic chambers at the sandy vegetated site in Barnegat Bay, June 1989. Cores were incubated at ambient Bay bottom light intensity (light), or in the dark (dark).