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Spatial and Temporal Variability of Surface Chlorophyll, Primary Production, and Benthic Metabolism in Rhode Island and Block Island Sounds

for the Rhode Island Ocean Special Area Management Plan 2010

by

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Executive Summary

The ecosystems of Rhode Island Sound (RIS) and Block Island Sound (BIS) are dependent on the growth of phytoplankton – single celled microscopic plants. It is the phytoplankton that ultimately support the growth of all of the animals in the sounds. Because of their importance, managing the sounds should include a consideration of where and when the phytoplankton are found and the rates at which they are growing in different areas throughout the annual cycle. Prior to the work described in this report, surprisingly little was known about the phytoplankton in RIS or BIS. In this study we were very fortunate to be able to work with local fishermen who agreed to collaborate with us and other members of the OSAMP science team to collect surface water samples from all over the OSAMP study area in both sounds over a full year. These water samples were retrieved and brought to our laboratories at the Graduate School of Oceanography (GSO) where they were filtered. The phytoplankton were collected on the filter and the photosynthetic pigment they contain (chlorophyll) was extracted and measured. This is a standard way of quantifying the amount of phytoplankton present in marine waters and is much less time consuming that counting the individual cells. Our results showed that during most of the year phytoplankton are significantly more abundant in the surface water of RIS then in BIS. The exception is during summer, when the weaker tidal currents in RIS allow the water column to become thermally stratified – that is, the surface waters are sufficiently warmer than the bottom water that the water column does not mix vertically. When the water column is not well mixed, the inorganic nutrients the phytoplankton need to grow become depleted in the sunlit surface water while they accumulate in the dark bottom waters where light limitation prevents phytoplankton from growing. The nutrients (nitrogen and phosphorus and silica) accumulate in the bottom water because the phytoplankton sink out of the surface waters when they die or they sink as fecal pellets if they are eaten by small animals in the surface water. Much of the nitrogen and phosphorus and silica that the sinking organic mater contains is regenerated on the bottom by animals and bacteria living on and in the sediments.

The chlorophyll measurements also showed that there was a strong bloom or period of rapid growth in abundance of the phytoplankton in the sounds during fall. This fall bloom was not seen in similar measurements made by the GSO plankton monitoring program in mid Narragansett Bay. Aside from the fall bloom period, the concentrations of phytoplankton in the sounds are significantly lower than found in the mid bay. This does not necessarily mean that the total abundance of phytoplankton beneath a given area of water in the sounds is lower than in the bay

because the sounds are much deeper and the water there is clearer than in the bay. Thus, phytoplankton may be able to live and grow at greater depths in the sounds. We began making measurements of how fast the phytoplankton were growing about six months after the chlorophyll measurements began when some extra funding became available. These measurements are much more technically challenging and we are still analyzing the measurements that have been made. It seems likely at this point that rates of growth will be similar to those measured by others in the lower West Passage on Narragansett Bay (100-200 g Carbon per square meter per year).

We also measured the rates at which organic matter was being consumed and nutrients were being regenerated on the bottom at stations in each sound where organic matter accumulated (areas with fine grained sediments relatively high in organic matter in contrast to the coarser sandy sediments that cover a larger part of the sounds). These measurements were made by dropping a large (0.25 m^2) box corer to the bottom and collecting sediments that were then brought up to the surface and put on the deck of a fishing boat. Smaller cores of this sediment were collected with plastic pipes and returned to the laboratory where they were kept in the dark at constant temperature (the same as measured in the bottom water in the sounds) in large walk-in environmental chambers. Filtered bottom water was carefully placed over the sediments in each core and changes in the concentrations of dissolved oxygen, ammonium, nitrite, nitrate, phosphate, and silicate were measured over many hours. These gave us a measure of how fast organic matter was being respired by the bottom and how fast nutrients were being regenerated. The rates appear similar in both sounds and appear to be significantly higher that similar rates in Narragansett Bay. This is probably due to the fact that the fine grained silt-clay sediment areas we were sampling in the sounds concentrate organic matter sinking to the bottom. All of these studies will be continued with support from RI Sea Grant and the National Science Foundation even though the OSAMP research program has concluded.

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Abstract

The goal of this project was to obtain the first measurements of phytoplankton biomass (standing crop) measured as chlorophyll over an annual cycle throughout Rhode Island and Block Island Sounds as well as the first measurements over an annual cycle of carbon fixation by the phytoplankton using modern ^{14}C uptake techniques. We also sought to obtain the first measurements of benthic-pelagic coupling in the sounds by incubating fine grained silt-clay sediment cores collected from two stations in Rhode Island Sound (RIS) and one in Block Island Sound (BIS). During these incubations we measured the sediment-water flux of dissolved oxygen and major inorganic nutrients (ammonium, nitrite, nitrate, phosphate, silicate). The first goal was achieved through collaboration with commercial fishermen and fellow OMSAP scientists who collected surface water samples many times at dozens of locations for our analyses. The seasonal cycle throughout both sounds was marked by a strong fall bloom that was not observed at a long-term monitoring station in mid Narragansett Bay. The bloom appeared to be synchronous throughout inner and outer reaches of both sounds. Chlorophyll concentrations in the surface water were lower than found in the mid bay. While surface chlorophyll was lower in RIS when it was thermally stratified during summer (1.3 $\mu g\ L^{-1}$ in RIS vs 2.0 $\mu g\ L^{-1}$ in BIS), the mean annual concentration was higher in RIS (2.7 $\mu g\ L^{-1}$ in RIS vs 1.8 $\mu g\ L^{-1}$ in BIS). It is possible that the higher mean annual chlorophyll in RIS reflects a higher annual carbon fixation that contributes to the higher biomass of fish and lobsters per unit area that was found in RIS by the National Marine Fisheries Service/Virginia Institute of Marine Science Northeast Area Monitoring and Assessment Program (NEAMAP) Near-Shore Trawl Survey. ^{14}C measurements of primary production by the phytoplankton became possible when additional funding became available, so only seven months of data have been acquired at this writing. Additional sampling is planned to complete the annual cycle. Based on the measurements in hand, we anticipate that annual production may be in the 100 to 200 g C $m^{-2}\ y^{-1}$ range in both sounds. This is similar to values measured previously by others in the lower West Passage of Narragansett Bay. While nutrient exchanges between the sediments and the overlying water are still being analyzed, the oxygen uptake data show little difference between the sounds. The oxygen uptake rates are significantly higher than measured in Narragansett Bay during the 1970s and 1980s, and are much higher than recent measurements in the mid bay. It is probable that these higher rates reflect the focusing of organic deposition in the fine-grained sediment areas where cores from the sounds were collected.

1 Introduction and Background

Marine ecology became a quantitative science in the late 1800s and early 1900s in order to answer a basic and very practical question, "why do some lakes or areas of the sea consistently yield greater catches than others?" For marine fisheries, the pursuit of this question led to the formation of the Kiel Commission for the Scientific Study of the German Seas, precursor to the current International Council for the Exploration of the Sea or ICES (Nixon et al. 1986, Mills 1989). Advances in analytical chemistry made it possible to begin to obtain measurements of the low concentrations of primary plant nutrients dissolved in sea water and the development of metered nets and coring devices made it possible to take quantitative samples of plants and animals in the water column and in sediments. The application of these tools led early researchers to see links between nutrient levels, phytoplankton (or, in shallower systems, macrophytes like eelgrass), zooplankton, and fisheries yields. As Karl Brandt, one of the leading marine scientists of the time, put it in 1901:

> The animals which inhabit the sea are developed in proportion to the quantity of their food. Now, since all this food comes directly or indirectly from plants, it follows that we can just as well estimate the real production of animal life in water by means of the annual yield of vegetation as we can estimate the product of a farm by the quantity of grass and fodder that it affords.

This view predominated for many decades and supported what the great fisheries ecologist D. H. Cushing (1975) called the "agricultural model" of marine productivity. But, of course, agriculture is necessary precisely because unmanaged nature does not often provide simple, low–diversity ecosystems with short, linear food chains that support high yields.

The development and application of radioactive (e.g. ^{14}C, ^{32}P, ^{3}H) and stable (e.g. $\delta^{13}C$, $\delta^{15}N$) isotopic techniques following the Second World War, as well as greatly enhanced optical and electron microscope capabilities, allowed us to see that measurements of standing crops of larger organisms missed a great deal of microbial complexity and a very dynamic cycling of nutrients in marine systems. The ways in which traditional grazing food chains leading to the production of relatively large and long-lived animals interact with the diverse microbial parts of the

ecosystem that turn over very rapidly remains a key question in marine ecology. And, of course, these interactions must respond in complex ways to changes over virtually all time scales in the physical environment.

In spite of all this complexity, it is still possible to see some first order correlations between primary production by phytoplankton and marine fish production (Iverson 1990) and harvest (Nixon 1988), at least in systems where hypoxia and over harvesting are not important stresses (e.g. Breitburg et al. 2009). Because there is a correlation between phytoplankton standing crop (measured as chlorophyll) and carbon fixation measured using ^{14}C uptake (e.g. various BZI models, Cloern et al. 1995, Brush et al. 2002), it is also possible in some cases to find correlations between phytoplankton chlorophyll and marine fishery yields (e.g. Ware and Thomson 2005). Because of these cross system correlations there is reason to believe that factors which influence the standing crop of phytoplankton may also influence the production of the phytoplankton and thus the productivity of fish, shell fish, and decapods (e.g. Nixon et al. 2009). Gaining some basic understanding of phytoplankton standing crop and productivity and the nutrient cycling that support this primary production is a first step in understanding and managing coastal marine fisheries ecosystems like Rhode Island and Block Island Sounds. The purpose of this project was to take this first step for the longer term Ocean Special Area Management Planning in Rhode Island.

In southern New England coastal waters there is strong stoichiometric as well as experimental evidence that the supply of dissolved inorganic nitrogen (DIN) is one of the most important factors that limits the standing crop and productivity of phytoplankton, especially during summer (e.g. Kremer and Nixon 1976, Oviatt et al. 1995, Smayda and Borkman 2008). And in inner shelf systems like Rhode Island Sound (RIS) and Block Island Sound (BIS), Rhode Island's major fishery ecosystems and the setting for the work described here (Fig. 1), the supply of DIN in the surface water is believed to be strongly regulated by the degree of vertical density stratification. This, in turn, is influenced by fresh water runoff, solar heating, wind mixing, and tidal stirring. The overall interplay of these factors appears to result in Rhode Island Sound being stratified during the warmer months and the more energetic Block Island Sound remaining generally well-mixed vertically year around (Shonting and Cook 1970, Edwards et al. 2004, Granger and Nixon unpublished survey data, Fig. 1). The transition in stratification between the two adjacent Sounds appears to be largely due to tidal mixing which can be seen in context using contours of the mean

Vertical Stratification Index (Simpson and Hunter 1974) calculated by C. Chen (U. Mass. Dartmouth, personal communication) using the FVCOM model (Fig. 2) (Chen et al. 2003).

Based on this difference in the physics of the two Sounds, we hypothesized that phytoplankton chlorophyll (and primary production per unit area) in smaller, well– mixed BIS (~ 635 km2, mean depth of 28 m) would be higher than in the much larger and seasonally stratified RIS (~ 1420 km^2, mean depth of 29 m). The reason being that deep DIN–rich shelf water coming into the Sounds will be more rapidly mixed up into the euphotic surface water in BIS as will DIN released from the bottom sediments as organic matter sinking out of the water column is decomposed. In other words, the productive pelagic part of the ecosystem is more tightly coupled with the heterotrophic regenerative bottom part in BIS than in RIS. On the other hand, RIS lies immediately off Narragansett Bay which receives large amounts of nitrogen from land drainage and direct sewage discharges (Nixon et al.2008). The importance of this for productivity in RIS is unknown. The concentrations of DIN in lower Narragansett Bay are about 2-4μM during the warmer months (Pilson 1985), but the volume of RIS is about 20 times that of the bay. BIS lies between RIS and Long Island Sound, and the latter also receives large amounts of land drainage. But water from Long Island Sound largely leaves the sound around Montauk Pt. at the tip of Long Island and may not mix much with BIS water (Edwards et al. 2004, D. Ullman, URI, personal communication). Hypoxia does not appear to be a significant factor in RIS or BIS.

It may seem surprising that we could not make a definitive statement about the comparative standing crop of phytoplankton and primary production in RIS and BIS prior to this study, especially given the wealth of marine research institutions surrounding them and their importance as fishery ecosystems. But these inner shelf coastal systems appear to have previously "slipped through the crack," perhaps because they are too small for blue water oceanographic research ships and too large and exposed for small inshore research vessels. In any case, we began the first systematic sampling program for chlorophyll (and light attenuation in the water column) in both sounds in December, 2008 and the sampling is on-going. Prior to this very few observations of water column ecology had been made in the systems (or at least published) since the pioneering studies by Gordon Riley in BIS over half a century ago (Riley 1952 a, b). At that time Riley (1952b) concluded, "… there seems to be little doubt that B.I.S. has a higher concentration of phytoplankton than the adjacent waters to the east and south…" An ongoing analysis of SeaWiFS sea surface color imagery by K. Hyde and J. O'Reilly at the National Marine Fisheries Service (NMFS) laboratory in Narragansett (personal communication)

suggests that the surface chlorophyll between 1998 and 2007 may have averaged about 2.1 mg m^{-3} in BIS compared with 1.4 mg m^{-3} in RIS. But this is based on an algorithm that has not been calibrated with local measurements of chlorophyll and primary production in the sounds. We are working with the NMFS scientists to develop a seasonally adjusted (if necessary) field verified algorithm for each sound so that we can make spatially explicit estimates of chlorophyll and productivity from satellite color imagery (SeaWiFS and MODIS). When additional OSAMP funding became available, C. Oviatt and J. Mercer began measurements of ^{14}C uptake by the phytoplankton at one station in RIS and the BIS station (along with supporting measurements of vertical light attenuation, temperature, and salinity) beginning in October 2009. Their work is also on-going. Methods for all of this analytical work are described in more detail in the methods sections below.

Strong benthic-pelagic coupling is characteristic of coastal systems. To document this connection, wanted to obtain the first measurements of the benthic metabolism in the sounds as measured by oxygen uptake and inorganic nutrient regeneration. Measuring the benthic metabolism would have the added benefit of providing a rough check on our estimates of primary production since cross-system comparisons have shown correlations between the primary production in the water and respiration on the bottom. As described above, our hypothesis was that nutrients regenerated on the bottom and in bottom waters would be more rapidly mixed up into the euphotic zone in RIS than in BIS and help to support higher primary production there.

2 Methods

2.1 Surface Chlorophyll and Vertical Light Attenuation

Surface water samples were collected in two ways. In the great majority of cases, bucket samples were collected, placed in opaque plastic bottles and kept cool in the dark until returned to the laboratory for processing. In a relatively few cases when primary production was also being measured, water was collected using a long tube as described below in the primary production methods section. The chlorophyll concentrations measured in the tube samples are not co-mingled with the concentrations from bucket samples. The largest number of samples were collected by two collaborating commercial fishermen during voyages of opportunity (Capt. Michael Marchetti and Capt. Rodman Sykes). While the time between water collection and

processing in the laboratory necessarily varied by station and circumstance, it was usually about five hours. Samples were also collected by collaborating OSAMP scientists and/or their students (D. Ullman, D. Codiga, J. King, P. Payton, S. McWilliams) in addition to our own laboratory personnel. Because voyages of opportunity were used as the only practical way in which to sample the large areas involved, surface chlorophyll samples were not collected on a fixed time scale or at fixed locations. In all cases, sample locations were taken using GPS.

Vertical light attenuation coefficients (-k m^{-1}) were calculated from profiles of light at various depths as described below. These measurements were made by our laboratory personnel or by collaborating scientists noted above. The commercial fishermen took Secchi disk readings (on a downcast) using a calibrated line and a 20 cm dia black and white disk. These were converted to approximate –k values using 1.7/D (Idso and Gilbert 1974). When available, -k values were also calculated from measurements of light attenuation with depth taken with a SeaCat 19 SeaBird Electronic equipped with a LiCor 4π light sensor.

From each sample bottle, three 100 ml aliquots of sample were filtered onto 0.7 μm GF/F filters (Whatman, 2.5 cm dia; Aminot and Rey 2000). Chlorophyll was extracted by placing each filter in 10 mL of 90% buffered acetone (20 ml for tube samples) for 24 hours (Cohen 1995). Following extraction, each sample was centrifuged for 5 minutes. Each extract was transferred to a clean 50 mL glass tube where it was well mixed before approximately 8 mL were transferred to a clean glass tube that was inserted into the fluorometer. Fluorescence readings were measured with a Turner Designs 10AU Laboratory Fluorometer.

Chlorophyll and phaeophytin concentrations (μgL^{-1}) were calculated using the EPA Method 445.0 by Arar and Collins (1997). After the initial fluorescence reading (f_o), two drops of 10% HCl were added and a second reading was taken (f_a).

$$\text{Chlorophyll } a \text{ (g/L)} = Fs * [r/(r-1)] * (fo-fa) * (V_e/V_f)$$

$$\text{Phaeophytin (g/L)} = Fs * [r/(r-1)] * (r*fa-fo) * (V_e / V_f)$$

Where:

Fs = known chlorophyll concentration / fluorometer reading
r = fo of pure chlorophyll standard / fa of pure chlorophyll standard
fo = fluorescence of sample, before acidification

fa = fluorescence of sample, after acidification
V_e = Volume of extracted chlorophyll *a*
V_f = Volume filtered

Mean chlorophyll and phaeophytin values were calculated from triplicate sub samples. Chlorophyll a concentrations were plotted versus daily areal productivity measured at the same time to determine if chlorophyll a concentrations could be used as a reliable indicator of primary production.

2.2 Primary Production

2.2.1 Sample Collection

In order to obtain estimates of primary production in the offshore waters of Rhode Island two sites were sampled; one in Block Island Sound and one in Rhode Island Sound (Fig 3). The Rhode Island Sound sampling site was approximately 15.5 kilometers northeast of the northern tip of Block Island at 71° 23.825' W and 41° 16.361' N. The depth at this station was approximately 40 meters. The Rhode Island sampling site was 7.0 kilometers southwest of the northern tip of Block Island at 71° 39.652' W and 41° 13.007' N. The depth at this station was approximately 36 meters.

When possible, water samples were collected by two different methods. The first method was to collect water using a Niskin bottle 1 meter below the surface. The second method involved using a 17 meter long by 3/4 inch diameter hose to obtain a depth integrated water sample. A hose with an open valve at the top end was lowered to 17 meters (~ avg. euphotic depth) at which point the valve was closed. The hose was then brought to the surface at which point the valve was again opened and the water drained into a large bucket and homogenized. This procedure is analogous to putting your thumb over the top of a straw in a glass of water and then removing the straw and the water it contains from the glass. With both sampling methods the water was filtered through a 300 µm-mesh screen (to remove large zooplankton) into opaque 1-L polyethylene bottles. The samples were then placed in a cooler filled with ambient seawater (to maintain ambient temperature) and transferred to the URI MERL laboratory. In addition, vertical profiles of temperature, salinity, PAR, and in situ fluorescence measurements were collected using a SeaBird CTD equipped with a Turner SCUFA submersible fluorometer and a Biospherical Scalar PAR sensor.

In order to obtain as many samples as possible, water was collected by two volunteer fishermen who collected samples when a University chartered boat was not available. These fishermen collected water samples using a Niskin bottle as noted above and also recorded Secchi depth measurements to be used in primary production calculations.

2.2.2 Primary Productivity by ^{14}C

Primary production was measured using a small volume/short incubation time method (Lewis and Smith, 1983) using standard ^{14}C procedures from Strickland and Parsons (1972). 20 mL borosilicate vials were spiked with 100 µL of 10 µCi/mL (1µCi for 5 mL of water) carbon-14 (^{14}C) stock solution. Individual water samples were gently mixed and 5 mL of sample was pipetted into the spiked 20 mL borosilicate vials. A total of 18 vials (16 light and 2 dark) were filled for each water sample, capped, and two vials were immediately placed into opaque covers. Each vial was placed into a specified location in an incubation tray.

Incubation trays were placed into a light and temperature controlled incubator and were incubated within 2°C of the *in situ* temperature for two hours. Each location in the tray had a specified light intensity (range 0-2000 $\mu Em^{-2}s^{-1}$) which was achieved by applying neutral-density screening to the underside of the tray. The light intensity at each vial location within the incubator was measured with a LiCor 192SA cosine irradiance sensor before the incubation period. The cosine values were converted to 4π readings using an empirically determined equation:

$$4\pi = 19.2 + 1.098\ (\cos) - 0.00011\ (\cos)^2$$

with both 4π and cosine light intensity in units of $\mu E\ m^{-2}\ sec^{-1}$. The light data measured in the incubator were converted prior to fitting P-I curves.

Upon removal from the incubator, 200 µL of 0.10N HCl was added to each vial, capped and mixed, and then uncapped again. Vials remain uncapped while gently agitated in the dark for approximately 40 hours to allow all of the unincorporated ^{14}C to be converted to CO_2 gas and removed from the sample. After this time period, 17 mL of MP Biomedicals Universol Scintillation Cocktail was added and the tightly capped vials were shaken vigorously. Measurements of ^{14}C converted into organic carbon are made on a Packard TriCarb Liquid

Scintillation Counter (Model 2900). The scintillation counter was configured to measure single labeled ^{14}C samples as disintegrations per minute (DPM) for five minutes.

To calculate the specific activity added on each incubation date, 100 µL of 10 µCi/mL ^{14}C stock was added to each of three vials containing 3 mL of β-phenylethylamine. After the stock was added, 17 mL of scintillation cocktail was added. The three specific activity vials, along with one blank containing 17 mL of scintillation cocktail, were counted with each set of samples.

2.2.3 Calculation of Primary Production

Volume specific primary production was calculated using equations similar to that of Strickland and Parsons (1972) as follows:

$$P(i) = \frac{(1.05DPM(i))DIC}{A_{sp}T}$$

$$P(d) = \frac{(1.05DPM(d))DIC}{A_{sp}T}$$

$$A_{sp} = DPM(sa)-DPM(back)$$

$$P(I) = P(i) - P(d)$$

Where:

P(*i*) = primary production rate at light intensity I (µgC L^{-1} h^{-1} or mgC m^{-3} h^{-1})
P(d) = dark production, (µgC L^{-1} h^{-1} or mgC m^{-3} h^{-1})
DPM(*i*) = dpm of sample incubated at light intensity I
DPM(d) = dpm of dark incubated sample
DPM(back) = background dpm in vial containing only scintillation cocktail
DPM(sa) = specific activity added to incubation samples (DPM)
T = incubation time (h)
DIC=concentration of dissolved inorganic carbon (µg/mL) (methods below)

For each of the water samples, a P – I curve (P(I) vs. the irradiance (I, µE m^{-2} s^{-1}) to which the incubating sample was exposed) was calculated from the data. The P – I curves were fit via one of two possible models, depending on whether significant photoinhibition occurred. In cases where photoinhibition was evident, the model of Platt *et al*. (1980) was fit to obtain the theoretical maximum production and terms for light-dependent rise in production and degree of photoinhibition.

$$P(I) = P_{sb}(1-e^{-a})e^{-b}$$

Where:

P(I) = primary production at irradiance I, corrected for dark fixation (P(i)-P(d))
P_{sb} = theoretical maximum production without photoinhibition
a = αI/ P_{sb} where α is the initial slope the light dependent rise in production
b = βI/ P_{sb}, where β is a term relaying the degree of photoinhibition.

If it was not possible to converge upon a solution, an alternative model of Webb *et al.* (1974) was similarly fit to obtain the maximum production and the term for light-dependent rise in production.

$$P(I) = P_{max}(1-e^{a'})$$

Where:

P(*I*) = primary production at irradiance I corrected for dark fixation (P(i)-P(d))
P_{max} = light saturated maximum production
a' = αI/ P_{max}, where α is the initial slope the light-dependent rise in production

P_{max} and P_{sb} are not equivalent but they are mathematically related using the equation:

$$P_{max} = P_{sb}\ [a/(a+b)][b/(a+b)]b/a$$

2.2.4 Light vs. depth profiles

To obtain a numerical representation of the light field throughout the water column averaged CTD light profiles (0.25 m intervals) were fit to the standard irradiance vs. depth equation:

$$I_z=I_o e^{-kz}$$

Where:

I_z=light irradiance at depth Z
I_o=incident irradiance (Z=0)
k=extinction coefficient

For each station profile, an extinction coefficient (k) was determined by regressing $\ln(I_z/I_0)$ *vs.* depth. The slope of the regression line estimates k (m^{-1}). When no CTD light profiles were available Secchi depth (Z_{sd}) measurements were made and k was calculated using the relationship determined by Holmes (1970) where $k = 1.44 / Z_{sd}$

Incident light data

Incident light data were collected and recorded at 15 min intervals using a Kipp & Zonen, Pyranometer Model SP Lite located at the nearby National Climatic Data Center (NOAA-NESDIS) Observatory in Kingston, RI. The total incident light data were converted to PAR using an empirically determined equation:

PAR = 0.4363 (Total Irradiance)

These data are used as the photoperiod incident light (I_o) time series in the following calculations.

2.2.5 Calculation of Daily and Areal Primary Production

Given the best fit parameters (P_{sb} or P_{max}, a, b) of the P – I curves obtained for each of the water samples, the photoperiod incident light (I_o) time series, and the extinction coefficient, it is possible to compute daily volumetric and areal production for each sample. Daily production ($\mu gC\ L^{-1}\ d^{-1}$) is obtained by integration of the determined activity throughout the photoperiod. The primary assumption of the approach is that the P – I relationship obtained at the time of sample procurement (towards the middle of the photoperiod) will be representative of the majority of production occurring during the photoperiod, which should be the case. Areal production ($mgC\ m^{-2}\ d^{-1}$) is obtained by integration of daily volumetric production vs. depth down to the depth of the euphotic zone. P – I parameters and extinction coefficients from the nearest sampling date and the actual photoperiod incident light series were used in order to obtain estimates for daily areal production on days that samples were not taken.

2.2.6 Dissolved Inorganic Carbon (DIC)

Inorganic carbon was determined by the measurement of carbon dioxide released by acidification of a sample. The sample was siphoned with a small bore tube into a 40 mL glass vial so as to not introduce any gas bubbles then 2 drops (0.1 mL) of sodium azide were added for preservation. The vial was capped with a Teflon/silica septa, making sure no bubbles were present, and stored at 4°C until analysis. Duplicate samples were collected, from which three replicates were measured. The sample was automatically injected into the instrument, which was fitted with a 0.343 mL sample loop, followed by the addition of 0.5 mL of 5% phosphoric acid.

As the pH of the sample is lowered, carbonate and bicarbonate ions are converted to dissolved carbon dioxide. High purity nitrogen purges any carbon dioxide and carries it to the molecular seive trap where it is held at 25°C, trapped, and concentrated. When this is complete, the trap is rapidly heated to 200°C. A stream of gas desorbs the carbon dioxide from the trap and carries it into a non-dispersive infrared analyzer (NDIR) which was calibrated to directly display the mass of carbon dioxide detected. Concentration of DIC was calculated by dividing this mass by the sample volume.

2.2.7 Adjustment for Rhode Island Sound Fall Bloom

When productivity measurements were compared to a time series of chlorophyll measurements from Rhode Island Sound it became obvious that our sampling dates missed a significant fall bloom. Over a 23 day time period between 10/20/2009 – 11/11/2009 (during which [Chl*a*] exceeded 4.5 μgL^{-1}) productivity values were adjusted to attempt to correct for this sampling bias. The first correction method utilized the chlorophyll *a* vs. production relationship described above. Nine chlorophyll measurements were obtained during the 23 day time period and were used to adjust daily areal production values over the time period by using chlorophyll concentration values from the closest possible date. This method however does not take into account differences in light intensity during the 23 day period. The second method followed the procedures described in the "Calculation of Daily and Areal Primary Production" section above but utilized P_{sb}, a, b, and k parameters from the January 19, 2010 sampling date as the average chlorophyll concentration over the 23 day time period was nearly identical to chlorophyll values from the January sampling date. This method allowed for the incorporation of variations in light intensity.

2.3 Benthic Metabolism

2.3.1 Field Collection

Triplicate sediment cores were collected on three occasions (fall, winter, and spring) from two stations in Rhode Island Sound and one station in Block Island Sound (Fig 3). Collections occurred when bottom water temperatures were 17˚C (October 2009), 4˚C (January 2010), and 7˚C (May 2010). The stations are located in depositional areas with fine-grain sediments that were found by ground truthing available sediment type data (McMaster 1960; U.S. Geological

Survey) and using suggestions made by J. Boothroyd (URI Geology, pers. comm.) and local fishermen. Intact sediment cores were collected using a 0.25 m^2 box corer. Three PVC subcores (30.5 cm long with a 10 cm inner diameter) were pre-mounted in the box corer (Hopkinson et al. 2001) to minimize disturbance of the surface layer during the coring operation. Cores were capped, dug out of the box corer, and maintained on deck in a dark cooler filled with water and held at ambient temperature during transportation back to the laboratory. Near-bottom water used during incubations (Hopkinson et al 2001; Fulweiler 2007; Fulweiler and Nixon 2009) was collected using a hose and pump and then filtered on deck down to 0.2 μm (A. Giblin, pers. comm.).

2.3.2 Analytical Methods

Upon returning to the laboratory, the cores were placed in a water bath in a dark temperature-controlled (at mean *in situ* field temperature) walk-in environmental chamber at the URI Graduate School of Oceanography. Cores were left uncapped with air stones gently bubbling oxygen through the overlying water in each core overnight (Hopkinson et al. 2001, Fulweiler 2007) to maintain the cores at saturation level. Before incubations, the overlying water in each core was gently siphoned off and replaced with 0.2 μm filtered *in situ* water (Hopkinson and Smith 2005) to reduce the effect of water column respiration during incubations.

During incubations cores are fitted with gas-tight covers with attached magnetic stir bars that slowly and continuously stirred the overlying water throughout the incubation (Fulweiler 2007; Fulweiler and Nixon 2009) to prevent stratification (Hopkinson et al. 2001) while avoiding sediment resuspension (Renaud et al 2008). Dissolved oxygen concentrations were monitored in the overlying water throughout the incubations using a Hach HQ30 LDO probe. Dissolved oxygen levels were allowed to drop by at least 2 ppm, but incubations were stopped before the overlying water reached hypoxia (a drop below 3 ppm; Fulweiler 2007; Hopkinson et al. 2001).

2.3.3 Data Analysis

Sediment oxygen consumption was determined from a linear regression of oxygen concentration over incubation time (Clough et al. 2005; Renaud et al. 2008). Values were adjusted for the volume and area of the core to obtain net fluxes (m^{-2} h^{-1}; Nixon et al. 1980). The differences in sediment oxygen consumption between seasons (temperatures) and locations were

examined using ANOVA. Statistical analyses were done using JMP software (version 5.1, SAS Institute, Inc.).

3 Results

3.1 Surface Chlorophyll

At the time of this writing we have analyzed over 825 samples of bucket collected surface water from the almost 3500 km^2 OSAMP area (Fig 3). Because of its large size, we broke the area into four sub areas consisting of inner and outer RIS and BIS. During the annual cycle sampled there was a clear seasonal cycle in all four areas, the most remarkable feature of which was a strong fall phytoplankton bloom (Fig 4). This bloom was not seen in mid Narragansett Bay by the GSO plankton monitoring program. On the other hand, a strong winter-spring bloom was observed in the bay during 2009-2010 that was not observed offshore and summer chlorophyll levels were considerably higher in the mid bay than offshore (Fig 5). Mean ± SD surface chlorophyll by region and by season is given in Table 1. Because the data were not normally distributed (and were not made so by any simple manipulation), we analyzed the differences between the sounds and between seasons using non-parametric statistics in JMP. Since there were no statistically significant differences between the inner and outer RIS or BIS, we pooled the inner and outer data. The pooled data for RIS showed significant differences between mean fall, winter, and summer or spring (which were not significantly different from one another) (Kruskal-Wallis test, $p < 0.001$). The same analysis of BIS seasonal means showed that each season differed significantly from all the others ($p< 0.001$). Differences between the two sounds were significant during winter ($p < 0.0179$) and summer ($p < 0.001$), but not during spring or fall. Comparison of the annual means also showed that RIS was significantly higher than BIS ($p < 0.001$). Analysis of the frequency distribution of surface chlorophyll in the two sounds showed that values above 4.5 $\mu g\ L^{-1}$ were unusual, but much more common in RIS than in BIS (Fig 6). The most common concentration observed in either sound fell between 0.5 and 1 $\mu g\ L^{-1}$.

3.2 Vertical Light Attenuation

Estimates of $-k\ m^{-1}$ based on the Secchi disk observations were consistent with the less frequent measurements of light attenuation taken with the SeaCat CTD (Fig 7). Water clarity was generally more variable in RIS than in BIS and there was no evidence of a clear seasonal cycle in

either area (Fig 7). The latter suggests that chlorophyll contributes relatively little to the absorption of light in the water column compared with other suspended solids and colored dissolved organic matter. There is the suggestion of increasing water clarity in both sounds over the period of study. A linear regression of k over time revealed a significant slope ($p<0.0001$) of the regression in BIS, but not in RIS.

Before proceeding with statistical analyses of the data, we excluded the one very high spring value from BIS and the two outlying high values during fall in RIS. These points fell outside an ellipse encompassing 99% of the data. While the data from RIS were normally distributed, those for BIS were not, so we again compared systems and seasons using non parametric analyses. In both sounds, the mean light attenuation was lowest during spring (Kruskal-Wallis test, BIS $p=0.0002$, RIS $p=0.0111$), and the summer mean attenuation was significantly ($p < 0.001$) higher in BIS than in the other seasons (Table 2).

3.3 Primary Production

3.3.1 Rhode Island Sound Productivity

Utilizing the Niskin bottle sampling technique, total productivity over the 7 month period from October 1, 2009 – April 30, 2010 was estimated at 79 gCm^{-2} (Fig 8). The integrated hose sampling yielded lower estimates for this time period at 39 gCm^{-2} (Fig 9). Measured Daily Areal Productivity in Rhode Island Sound (RIS) ranged from 70 $mgCm^{-2}\ d^{-1}$ to 1116 $mgCm^{-2}\ d^{-1}$ (Table 3).

Measured productivity in RIS was lowest on the January 19, 2010 sampling. However, this was mainly due to the low amount of incident light on the date the samples were collected. When the parameters estimated from the incubations on this date were modeled with incident light from preceding and following days, the estimated production was relatively high (avg. Niskin 469 mgC $m^{-2}\ d^{-1}$, hose 373 mgC $m^{-2}\ d^{-1}$) compared to estimates throughout the rest of the winter months. This sampling date also corresponds to the highest chlorophyll a concentrations measured on productivity sampling dates (6.1µg l^{-1}). During this time period an intense winter/spring bloom occurred in Narragansett Bay (Oviatt, Monitoring Network data).

Niskin sampling of surface waters resulted in highest productivity estimates (1116 mgC $m^{-2}\ d^{-1}$) for Rhode Island Sound on April 6, 2010. This sampling date was just after large amounts of rain and record flooding in Rhode Island which lasted from March 30 – April 1. A

chlorophyll concentration spike in Narragansett Bay was also observed immediately following the flooding (MERL data). One possible reason why the total estimated production over 7 months of samples collected with the hose method were lower than the Niskin method was that samples utilizing the hose method were not available for this date. This sharp spike in production was short lived, with both Niskin and hose sample estimates decreasing to 324 and 233, respectively, by April 14, 2010.

When additional RIS chlorophyll measurements (Fields, Codiga, Ullman, SAMP data) were incorporated into the analysis it became clear that a fall bloom occurred in RIS and was slightly later than the one in BIS, beginning at the end of October and therefore missed by our sampling. We have used two corrections to account for this bloom. The first utilized the chlorophyll to production relationship obtained from all of the Niskin bottle samples (Fig 10). The second involved modeling using parameters from the January 19, 2010 sampling date when similar chlorophyll values were observed. When the corrections were made to the Niskin measurement and the productivity was summed over the 7 month period total productivity values of 86 gCm^{-2} from the modeled correction (Fig 11) and 91 gCm^{-2} from the chlorophyll relationship were obtained. This range is similar to the Niskin productivity measurements of 87 gCm^{-2} from BIS (see below).

3.3.2 Block Island Sound Productivity

Utilizing the Niskin bottle sampling technique, total productivity over the 7 month period from October 1, 2009 – April 30, 2010 was estimated at 87 gCm^{-2} (Fig 12). The integrated hose sampling yielded lower estimates for this time period at 78 gCm^{-2} (Fig 13). Measured Daily Areal Productivity in Block Island Sound (BIS) ranged from 59 mgC m^{-2} d^{-1} to 1738 mgC m^{-2} d^{-1} (Table 3). As for RIS the lowest measured productivity in BIS was observed on the January 19, 2010 sampling date due to the low incident light on that specific date. When parameters were applied to incident light values from preceding and following days the average productivity values obtained from the Niskin sampling suggested that productivity was highest for the winter months at this time. The hose sampling method indicated that productivity was slightly higher in February than January. Chlorophyll values supported these observations.

In BIS chlorophyll a concentrations were highest on the October sampling date with values of 8.3 μgl^{-1} obtained from Niskin sampling and 7.7 μgl^{-1} from integrated hose sampling. The productivity measurements obtained from this date reflected these high chlorophyll values

with measured values of 1738 mgC m^{-2} d^{-1}, much higher than values observed during the rest of the 7 month study period. The measured productivity from the hose sample was 563 mgC m^{-2} d^{-1} which was relatively high, yet when the parameters were applied to the incident light for the 36 closest days the average productivity over this time period was less than the productivity from January and February.

3.3.3 Niskin versus Hose Estimations of Primary Production

Except for two occasions surface Niskin samples resulted in higher estimates of primary production than integrated depth or hose samples (Table 3). Most of the active chlorophyll was likely present in surface waters of the Niskin sampling procedure. Samples recovered from over the euphotic depth resulted in lower concentrations of active chlorophyll and reduced primary production estimations. In January and February when higher productivities were measured over the euphotic depth than from surface samples sub surface chlorophyll concentrations from winter blooms may have been present. Surface samples produced the highest potential estimations of primary production and integrated depth samples produced lowest potential estimations of primary production.

3.4 Benthic Metabolism

Sediment cores were collected and incubated at three different *in situ* bottom water temperatures (17°C, 7°C, and 4°C) from three stations (1 in BIS, and 2 in RIS, Fig 3). Data were normally distributed, so all comparisons were made using ANOVA in JMP. Data from the RIS 2 station were logarithmically transformed to obtain a normal distribution for statistical analysis across incubation temperatures.

At all three stations, mean sediment oxygen uptake decreased with decreasing temperature (Fig 14). This difference was most dramatic in BIS, where the mean O_2 uptake values were significantly different for all three incubation temperatures ($p=0.0058$). In RIS 2 (the easternmost RIS station, Fig 3), only the O_2 uptake at the warmest temperature (17°C) was significantly different than the rest ($p=0.0006$). At the other RIS station (RIS 1, the westernmost RIS station), the differences in sediment oxygen uptake between temperatures was not statistically significant.

There was no clear trend in rates of sediment oxygen uptake between stations at different incubation temperatures (Fig 14). At the two cooler bottom water temperatures (7°C and 4°C), the RIS 1 station exhibited the highest rate of benthic metabolism ($p=0.0175$ for 7°C, and $p=0.005$ for 4°C). During the warmest incubation (17°C), the BIS station had the highest rate of O_2 uptake, although none of the differences between stations was statistically significant. Table 4 summarizes the statistically significant differences in mean benthic metabolism across temperatures and stations.

To compare benthic metabolism in BIS and RIS overall, we pooled the data from both RIS stations. Sediment core incubations for Block Island and Rhode Island Sound revealed no significant differences in overall mean benthic metabolism between the sounds over the 13°C temperature range. However, there was a significant difference in mean sediment oxygen uptake (mg m^{-2} h^{-1}) for both sounds between each of the three incubation temperatures (one-way ANOVA, $p=0.0001$). For the two warmer bottom-water temperatures (17°C and 7°C), mean oxygen uptake in BIS was greater than in RIS although the differences were not statistically significant (Table 5).

4 Discussion

It is, of course, inappropriate to generalize too widely based on the relatively short sampling reported here (one year for chlorophyll and light attenuation, seven months for primary production, and three benthic flux measurements, albeit it across a wide temperature range). Based on the results so far, it appears that the seasonal pattern in phytoplankton abundance in the sounds is consistent in terms of timing across both RIS and BIS and in both inner and outer sound areas. The seasonal cycle in the sounds was quite different from that observed over the same time period in Narragansett Bay. Surface chlorophyll concentrations in the sounds were, as expected, considerably lower than found in mid Narragansett Bay. Our initial hypothesis that standing crops of phytoplankton would be higher in relatively well-mixed BIS than in RIS does not appear to be correct, except during summer when RIS is thermally stratified. On an annual basis, the opposite appears to be the case. This also contrasts with Riley's (1952b) belief based on cell count data from over half a century ago and the preliminary satellite sea surface color analysis of decadal annual mean chlorophyll (Hyde and O'Reilly,NMFS Narragansett, personal communication). The absolute values of annual means estimated from the satellite data (for years

prior to the sampling reported here) are not very different from our measured annual means (RIS 2.7 ± 2.2 our data vs 1.4 μg L^{-1} satellite; BIS 1.8 ± 1.4 our data vs 2.1 μg L^{-1} satellite). It was also surprising to us that the vertical light attenuation in both systems showed so little evidence of a seasonal cycle and the differences between the sounds were not statistically significant except during summer. Clearly, the thermal stratification in RIS results in significant differences in the base of the food chain in the two sounds during summer.

While the higher mean annual surface chlorophyll in RIS may well mean that annual primary production is higher there as well, we are not yet able to assess that directly with the measurements in hand. For the measurements made so far, there was little difference between integrated seven month production of RIS (79 g C m^{-2}) and BIS (87 g C m^{-2}) based on surface water only. As noted in results, tube samples of the euphotic zone yielded much lower values in RIS (39 g C m^{-2}) and slightly lower values in BIS (78 g C m^{-2}). We are still working to reconcile the apparent differences and to acquire a full annual cycle of sampling so that we do not have to make numerous assumptions about how to handle the production during the large fall bloom that occurred before the present sampling program began.

We began the introduction to this report by noting the cross system studies showing correlations between phytoplankton standing crops, primary production, and the yields of fish. While fisheries landings data are not reported separately for the sounds, a great benefit of the OSAMP process was that it stimulated interactions between the local research community and the recently completed Northeast Area Monitoring and Assessment Program (NEAMAP) Near-Shore Trawl Survey (Bonzek et al. 2009). This NMFS funded program carried out by the Virginia Institute of Marine Science (VIMS) sampled 17 stations in RIS and 10 stations in BIS using standard trawls during spring and fall. The principal investigator of that project was kind enough to provide us with average numbers and biomass for each species per standard trawl in RIS and BIS between fall 2007 and spring 2009 (C. Bonzak, personal communication, Table 6). The result was that the total number and biomass of organisms captured per unit effort was greater during both spring and fall sampling in RIS than in BIS. This was true for pelagic as well as demersal species and almost always the case for all of the most abundant and commercially important species (Table 6). Since fishing pressure appears to be similar in both systems and both are subject to the same fisheries management regulations, this difference may reflect a great productivity in RIS, consistent with the surface chlorophyll data.

The benthic respiration data collected thus far did not indicate large differences between oxygen uptake by the fine grained sediments in the two sounds. However, benthic flux data are notoriously variable and additional sampling will be carried out with support from other sources to make measurements throughout an annual cycle. Since most of the bottom in both sounds is composed of coarser grained sediments than the depositional areas sampled here, we will also be sampling those areas, though we anticipate that the metabolic rates will be significantly lower. For both BIS and RIS at all incubation temperatures, mean oxygen uptake was much higher than recent measurements made in nearby Narragansett Bay (Fig 15, Fulweiler 2007). In every case, measurements in BIS and RIS were higher than historic Narragansett Bay values of benthic metabolism from Nixon et al (1976, 1980, unpublished data). Since the primary production data collected thus far do not suggest that depth integrated primary production in the sounds is higher than it was historically in the bay (Nixon et al. 2009), the higher rates of benthic metabolism in the fine grained sediments in the sounds probably reflect the fact that they are depositional areas in which sinking organic matter accumulates.

5 Conclusions

• Both Rhode Island Sound (RIS) and Block Island Sound (BIS) appear to be productive inner shelf systems where primary production may ultimately be found to fall between 100 and 200 g C m-2 y-1, though this must await the completion of an annual cycle of measurements now underway.

• The concentration of phytoplankton in the sounds (measured as chlorophyll) was substantially lower than in mid Narragansett Bay except during fall, when the sounds experienced a marked bloom not seen in the bay.

• The concentration of phytoplankton in surface waters was higher in RIS than in BIS except during summer, when the thermally stratified RIS was less turbid and contained lower phytoplankton concentrations.

• Based on spring and fall sampling carried out by the Northeast Area Monitoring and Assessment Program (NEAMAP) Near-Shore Trawl Survey (Bonzek et al. 2009), it appears that the abundance and biomass of fish and lobsters per unit area is greater in RIS than BIS, consistent with the higher mean annual chlorophyll concentrations found in the surface water.

• Rate of oxygen uptake by the fine grained sediments in both sounds are higher than measured historically (1970s and 1980s) and recently in Narragansett Bay, perhaps reflecting the depositional nature of the areas where these sediments accumulate.

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Table 1. Overall and seasonal mean (± SD) surface chlorophyll listed by region.

Location	Area* (km^2)	n	Overall Mean Surface Chlorophyll ± SD ($\mu g\ L^{-1}$)	Seasonal Mean Surface Chlorophyll ± SD ($\mu g\ L^{-1}$)			
				Summer (Jun-Aug)	Fall (Sept – Nov)	Winter (Dec – Feb)	Spring (Mar – May)
Inner BIS	728	353	1.82 ±1.42	2.15 ±0.57	3.8 ±1.48	2.73 ±1.44	0.98 ±0.73
Inner RIS	784	213	2.78 ±1.83	1.72 ±0.80	4.41 ±1.92	2.86 ±1.23	1.12 ±0.87
Outer BIS	337	40	1.80 ±1.14	1.29 ±.38	3.81 ±1.23	2.30 ±0.97	0.85 ±0.37
Outer RIS	1,570	233	2.57 ±2.42	0.90 ±0.22	4.65 ±3.3	3.0 ±0.95	0.92 ±0.53
Total BIS	1,065	393	1.81 ±1.39	2.04 ±0.62	3.80 ±1.46	2.64 ±1.36	0.98 ±0.70
Total RIS	2,354	446	2.67 ±2.16	1.30 ±0.71	4.53 ±2.70	2.93 ±1.10	1.00 ±0.70

*Areas given here have been adjusted to include the ocean SAMP study area (McCann 2009), and are not the traditional literature values used when referencing these regions.

Table 2. Mean extinction coefficient (-k) values for Block Island and Rhode Island Sound overall and by season. Letters show statistically significant differences ($p<0.05$) based on Kruskal-Wallis test (values not connected by the same letter are significantly different).

Region	n	Overall Mean –k (m^{-1})	Seasonal Mean –k (m^{-1})			
			Summer (Jun-Aug)	Fall (Sept -Nov)	Winter (Dec – Feb)	Spring (Mar – May)
Block Island Sound	51	0.3 ± 0.06	0.35 ± 0.03 a	0.29 ± 0.04 c	0.31 ± 0.03 d	0.25 ± 0.06 cd
Rhode Island Sound	60	0.29 ± 0.08	0.31 ± 0.06 b	0.27 ± 0.07 bc	0.33 ± 0.1 d	0.21 ± 0.04 cd

Table 3. Results from primary production sampling and CTD water column profiles. k is the calculated extinction coefficient from which Zeu (euphotic depth) was calculated. Pmax/Psb, a, and b were determined by fitting either the Platt (Psb, a, b) or Webb model (Pmax, a) to the P vs. I relationship obtained from each productivity incubation. Model start and end dates are the period over which the parameters were applied when total productivity calculations were made using a daily time series of total incident light.

Cruise	Site	Method	Date	Model Start Date	Model End Date	Model Period (Days)	k	Zeu	P_{max} / P_{sb}	a	b	[Chl a] (μgL^{-1})	Measured Daily Areal Productivity ($mgCm^{-2}d^{-1}$)	Avg Daily Areal Productivity Over Period ($mgCm^{-2}d^{-1}$)	Stratified	Depth of pycnocline (m)
SP1002	RIS	Niskin	10/2/2009	10/1/2009	11/1/2009	33	0.155	29.8	2.263	0.019	0.0012	0.844	235.16	160.97	Y	24
SP1002	RIS	Hose	10/2/2009	10/1/2009	11/4/2009	35	0.155	29.8	0.494	0.007		0.913	70.27	49.52	Y	24
SP1002	BIS	Niskin	10/2/2009	10/1/2009	10/25/2009	26	0.341	13.5	37.289	0.308	0.0180	8.323	1737.92	1279.60	Y	21
SP1002	BIS	Hose	10/2/2009	10/1/2009	11/4/2009	36	0.341	13.5	11.213	0.110	0.0064	7.653	563.36	390.88	Y	21
SP1118	BIS	Niskin	11/18/2009	10/26/2009	12/19/2009	54	0.288	16	1.950E+08	0.040	3.397E+05	2.675	200.02	185.39		
SP1202	RIS	Niskin	12/2/2009	11/2/2009	12/26/2009	54	0.320	14.4	16.453	0.098	0.0106	3.901	386.88	340.43		
SP1207	RIS	Hose	12/7/2009	11/5/2009	12/28/2009	55	0.404	11.4	15.663	0.050	0.0131	3.948	132.85	149.39	N	
SP1208	BIS	Hose	12/8/2009	11/5/2009	12/29/2009	54	0.320	14.4	7.324	0.026	0.0062	1.821	132.67	95.63	Y	28
SP0119	RIS	Niskin	1/19/2010	12/27/2009	1/27/2010	33	0.320	14.4	14.828	0.149	0.0018	6.069	79.49	469.44	N	
SP0119	RIS	Hose	1/19/2010	12/29/2009	1/27/2010	30	0.320	14.4	2.460E+08	0.083	4.264E+05	6.075	46.77	373.05	N	
SP0119	BIS	Niskin	1/19/2010	12/20/2009	1/27/2010	40	0.314	14.7	29.594	0.104	0.0225	5.974	58.99	421.21	N	
SP0119	BIS	Hose	1/19/2010	12/30/2009	1/27/2010	30	0.314	14.7	13.208	0.163	0.0075	4.549	86.58	464.77	N	
SP0205	RIS	Niskin	2/5/2010	1/28/2010	2/21/2010	25	0.290	15.9	12.203	0.071	0.0009	4.301	454.27	446.65	N	
SP0205	RIS	Hose	2/5/2010	1/28/2010	3/11/2010	42	0.290	15.9	2.035E+05	0.023	278.447	3.771	185.16	175.26	N	
SP0205	BIS	Niskin	2/5/2010	1/28/2010	2/21/2010	25	0.355	13	14.513	0.097	0.0075	5.026	452.29	442.89	WEAK	
SP0205	BIS	Hose	2/5/2010	1/28/2010	3/11/2010	42	0.355	13	18.511	0.235	0.0041	4.802	857.82	813.53	WEAK	
SP0310	RIS	Niskin	3/10/2010	2/22/2010	3/23/2010	30	0.241	19.13	2.561	0.023		1.308	207.06	141.15	WEAK	
SP0310	BIS	Niskin	3/10/2010	2/22/2010	3/27/2010	34	0.239	19.32	3.816	0.011	0.0026	1.402	130.89	89.70	WEAK	
SP0406	RIS	Niskin	4/6/2010	3/24/2010	4/10/2010	17	0.206	22.4	17.633	0.099	0.0030	2.864	1115.60	1089.67		
SP0414	RIS	Niskin	4/14/2010	4/11/2010	4/22/2010	12	0.156	29.5	2.729	0.020	0.0002	0.884	392.43	324.27	Y	15
SP0414	RIS	Hose	4/14/2010	3/12/2010	4/22/2010	42	0.156	29.5	2.107	0.021	0.0006	2.486	334.64	232.89	Y	15
SP0414	BIS	Niskin	4/14/2010	3/28/2010	4/22/2010	25	0.216	21.3	5.577	0.024		1.332	448.73	322.33	WEAK	
SP0414	BIS	Hose	4/14/2010	3/12/2010	4/22/2010	42	0.216	21.3	9.596	0.015	0.0140	1.449	257.59	172.61	WEAK	
SP0430	RIS	Niskin	4/30/2010	4/23/2010	4/30/2010	8	0.227	20.3	2.864	0.025	0.0002	0.937	320.49	232.88	Y	24
SP0430	RIS	Hose	4/30/2010	4/23/2010	4/30/2010	8	0.227	20.3	3.609	0.020	0.0018	1.049	286.20	204.60	Y	24
SP0430	BIS	Niskin	4/30/2010	4/23/2010	4/30/2010	8	0.214	21.5	22.539	0.039	0.0157	2.103	826.74	555.52	WEAK	
SP0430	BIS	Hose	4/30/2010	4/23/2010	4/30/2010	8	0.214	21.5	9.880	0.030	0.0025	2.198	622.04	419.20	WEAK	

Table 4. Statistically significant differences in benthic metabolism across temperatures and core collection stations. Each letter represents a significant difference (stations and temperatures with the same letter are not significantly different from each other).

Temperature	Station		
	RIS 1	RIS 2	BIS
17°C	a	a	a
7°C	a	b	b
4°C	a	b	c

Table 5. Mean ± SD sediment oxygen uptake for Block Island and Rhode Island Sounds determined from three sediment core incubations at mean *in situ* bottom water temperatures of 17°C (Oct 2009), 7°C (May 2010), and 4°C (Jan 2010).

Sound	n	Mean O_2 uptake (mg m^{-2} h^{-1})			
		Overall	17°C	7°C	4°C
Block Island Sound	9	54.91 ±30.79	86.61 ±13.26	55.88 ±22.11	22.25 ±4.21
Rhode Island Sound	18	44.54 ±25.38	66.25 ±13.97	39.93 ±27.61	27.45 ±17.05

Table 6. Comparison of mean biomass (kg live weight per trawl) found in Rhode Island Sound (RIS) and Block Island Sound (BIS) during standard seasonal surveys carried out by the Virginia Institute of Marine Science (VIMS) for the NMFS. Data courtesy of Dr. Chris Bonzek, VIMS. Sampling details are given in Bonzek et al. (2009).

	Fall '07	Spring '08	Fall '08	Spring '09
		TOTAL BIOMASS		
RIS	271	262	431	292
BIS	161	129	190	200
		SCUP		
RIS	34	20	84	90
BIS	24	14	41	24
		DOGFISH		
RIS	4	20	104	58
BIS	6	7	4	24
		SQUID		
RIS	51	5	33	1
BIS	31	4	28	5
		WINTER FLOUNDER		
RIS	6	20	8	12
BIS	0.3	9	2	16
		TOTAL FOUNDER		
RIS	13	30	14	18
BIS	5	16	7	22
		TOTAL SKATES		
RIS	90	113	119	71
BIS	40	65	75	113
		LOBSTER		
RIS	3.2	4.4	4.7	4.9
BIS	0.4	0.9	0.2	1.1

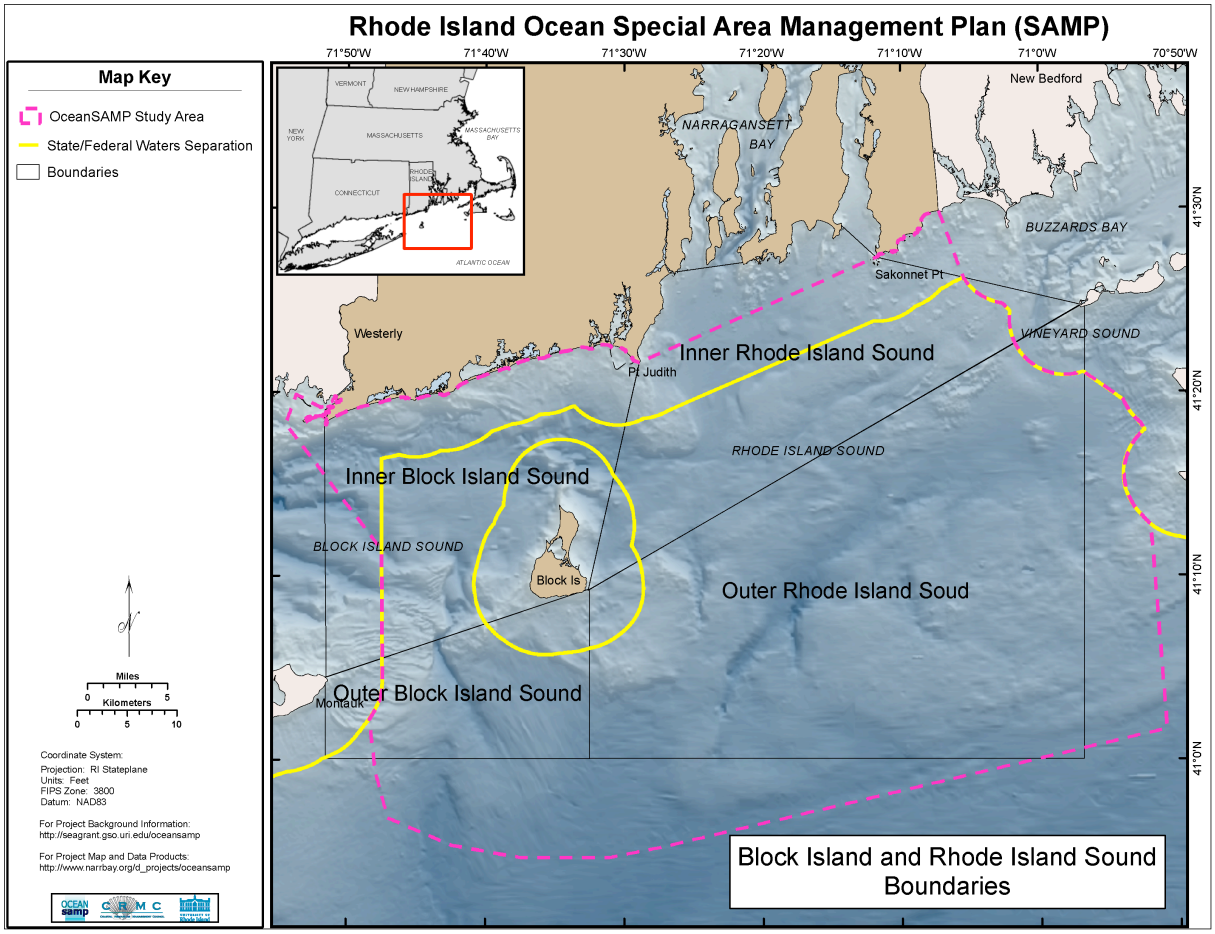

Fig 1. Ocean SAMP study area (McCann 2009) and delineations of inner and outer Block Island and Rhode Island Sound used in this study.

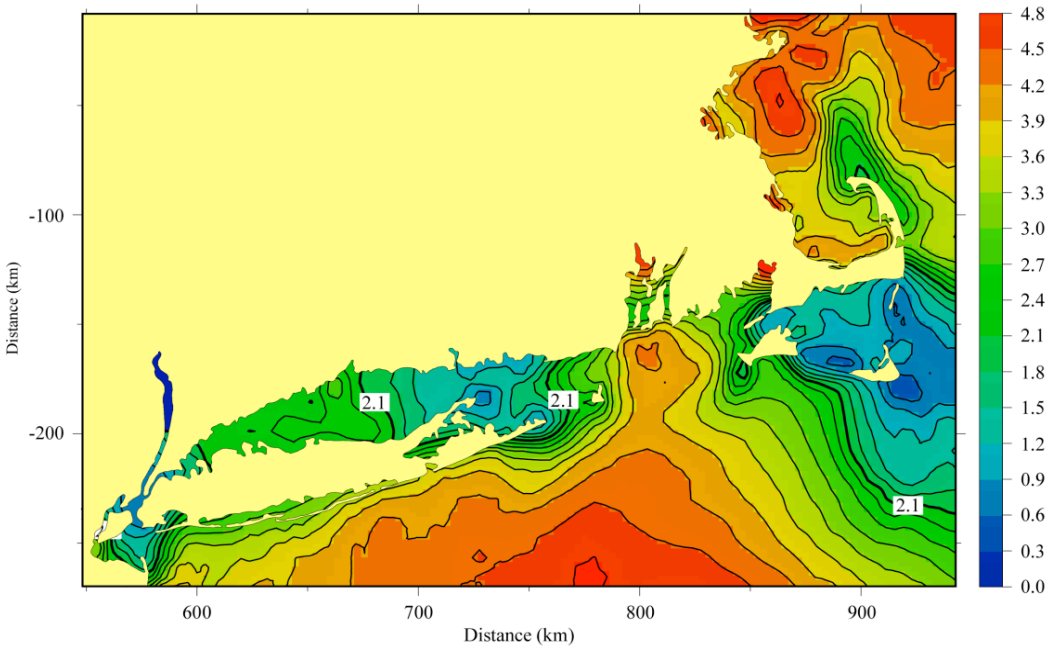

Fig 2. Contours of Simpson and Hunter's (1974) "Vertical Stratification Index" (log h/v^3) for southern New England coastal areas calculated by C. Chen (U. Mass. Dartmouth) using FVCOM model (Chen et al. 2003). Red-brown is strongly stratified while blue is well mixed.

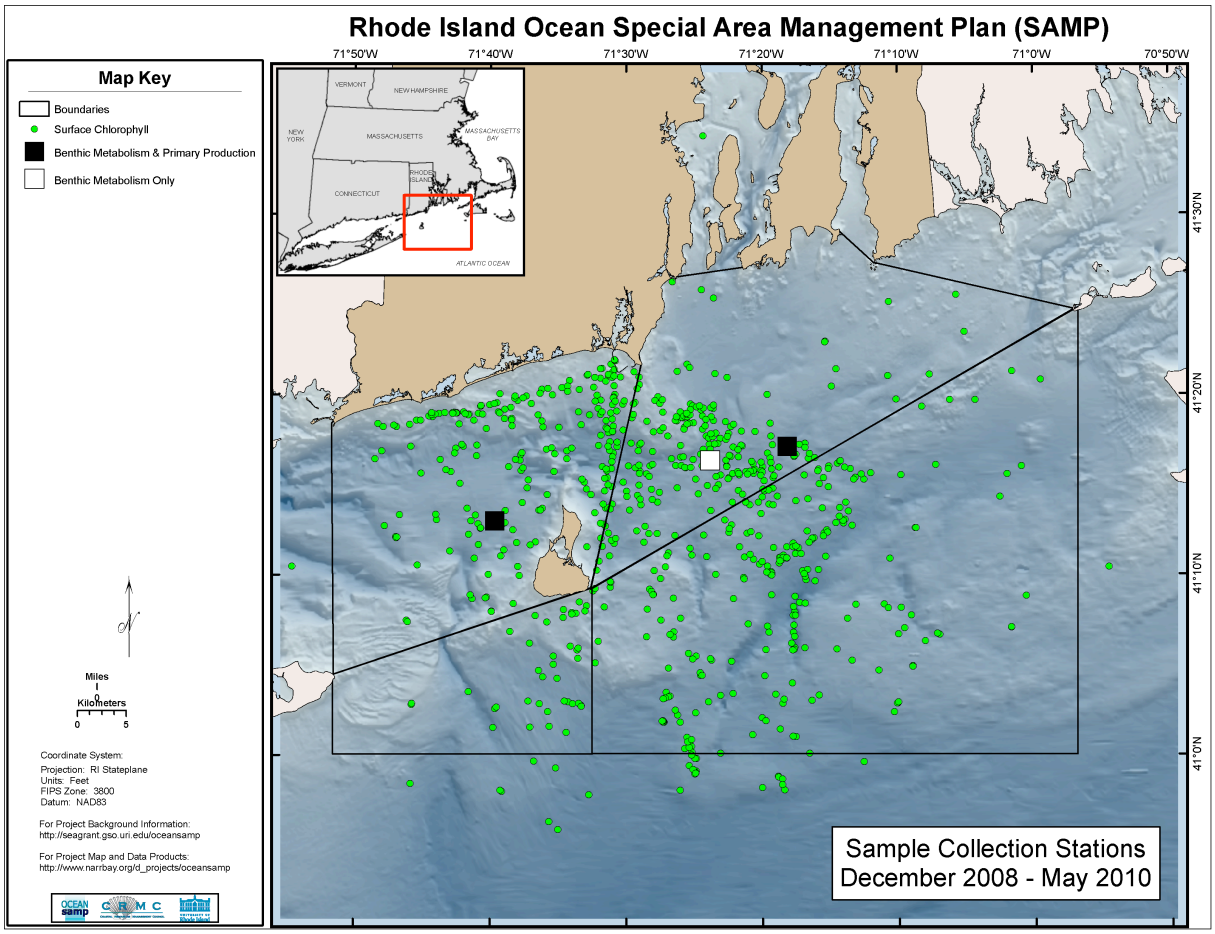

Fig 3. Surface chlorophyll, primary production, and benthic metabolism sampling stations in Block Island and Rhode Island Sound. Benthic metabolism stations are referred to as BIS (Block Island Sound station), RIS 1 (western Rhode Island Sound station), and RIS 2 (eastern Rhode Island Sound station).

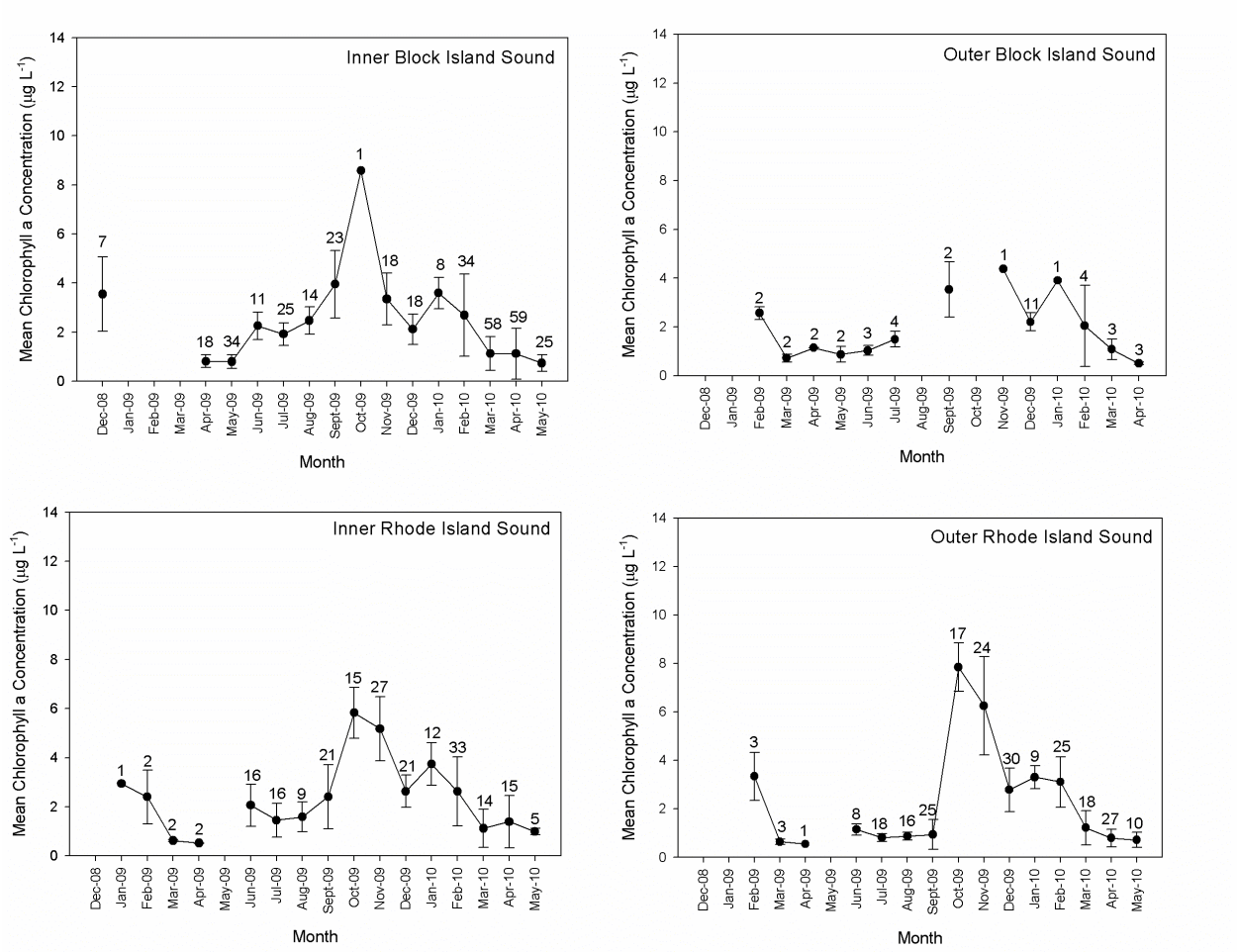

Fig 4. Monthly mean surface chlorophyll concentrations in (top left) inner BIS, (top right) outer BIS, (bottom left) inner RIS, and (bottom right) outer RIS. Error bars represent standard deviation, numbers above bars represent sample size.

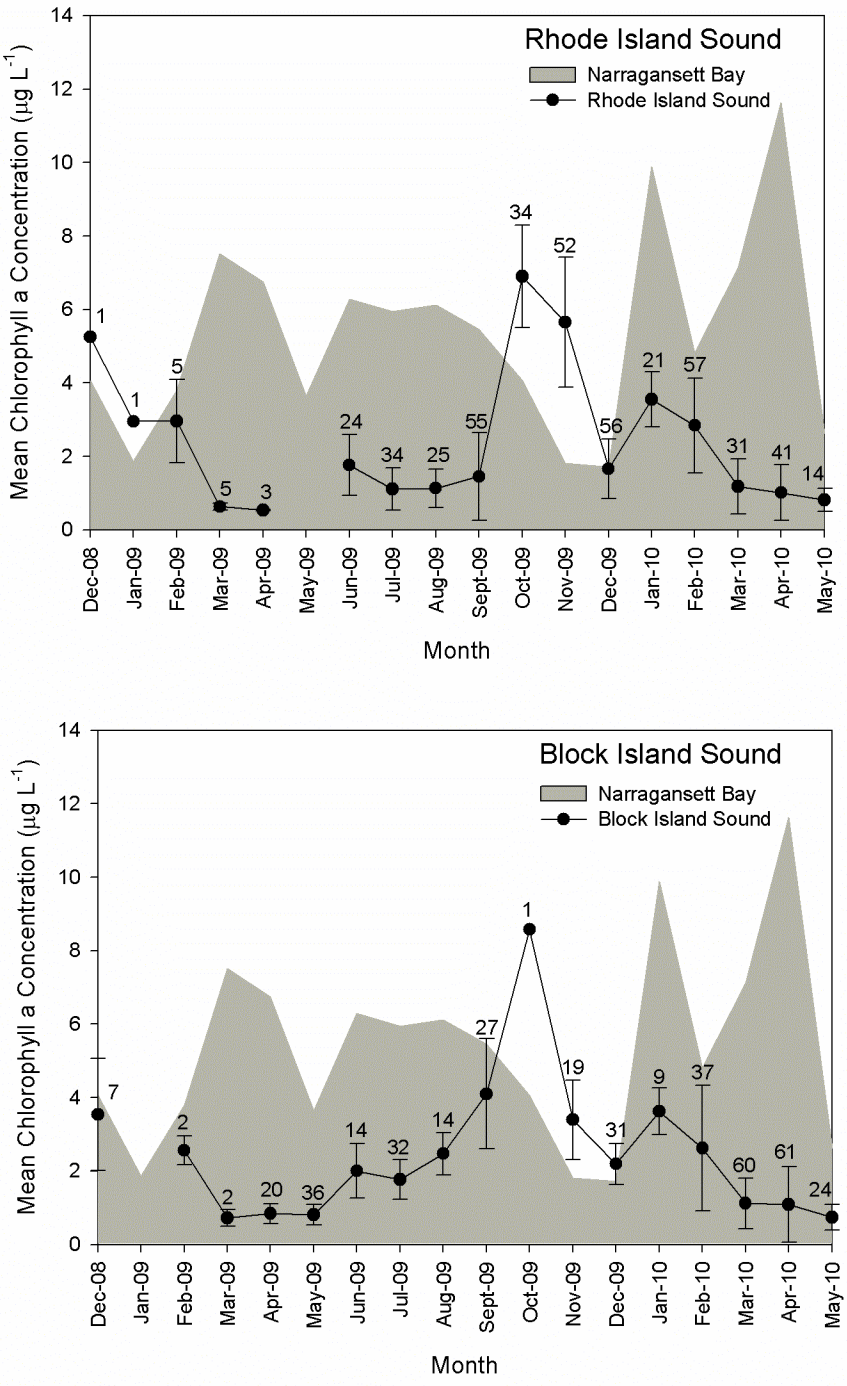

Fig 5. Monthly mean surface chlorophyll concentrations in (top) Rhode Island Sound and (bottom) Block Island Sound. Error bars represent standard deviation, numbers above bars represent sample size. Shaded areas are monthly mean chlorophyll concentrations calculated from the weekly sampling of the URI Graduate School of Oceanography long-term monitoring program (www.gso.uri.edu/phytoplankton).

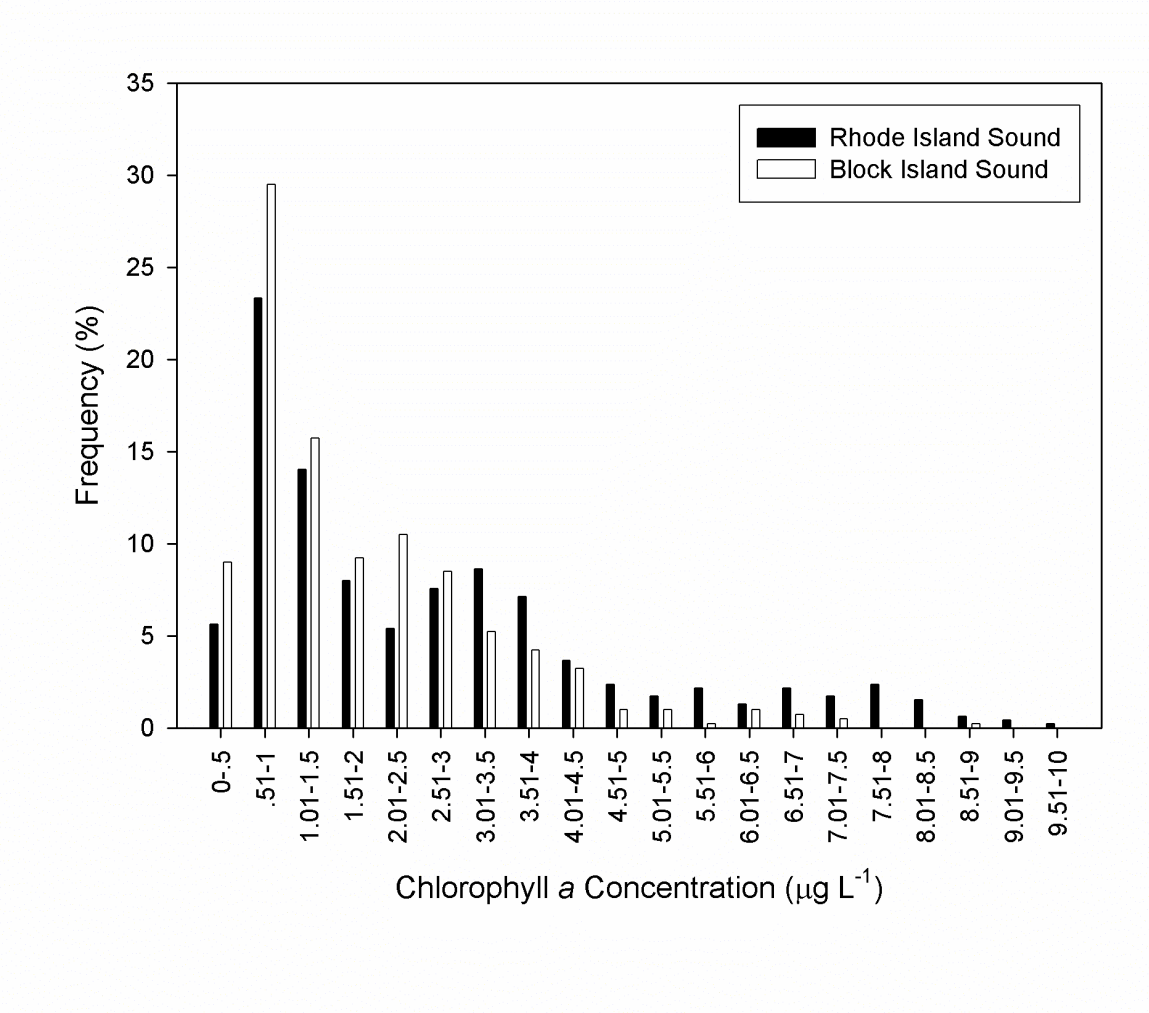

Fig 6. Frequency of surface chlorophyll values measured in Block Island and Rhode Island Sounds, binned in 0.5 μg L^{-1} increments.

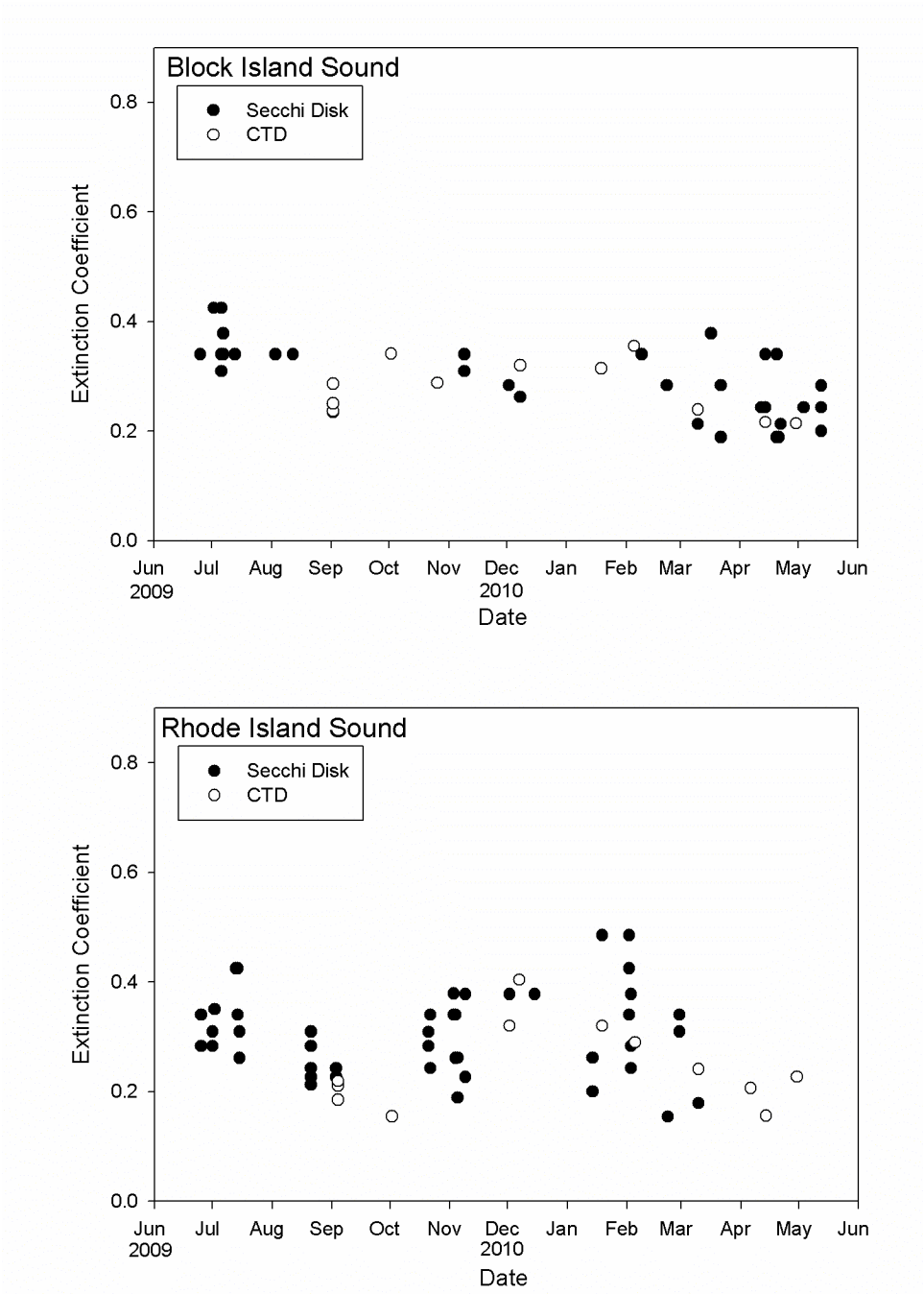

Fig 7. Extinction coefficients (k, in m^{-1}) plotted over time for (top) Block Island and (bottom) Rhode Island Sounds. K values were calculated from Secchi disk (opaque circles; Idso and Gilbert 1974) and CTD light profiles (open circles).

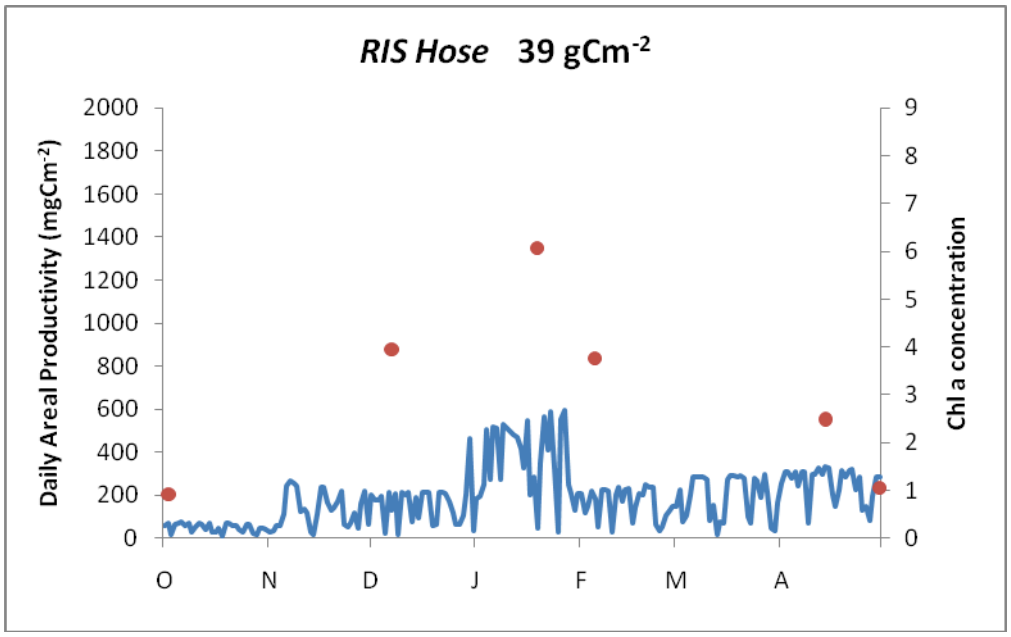

Fig 8. Daily Areal Production (blue lines) estimates and measured chlorophyll a concentrations (red dots) from RIS utiizing hose sampling method. The dates corresponding to the chl a concentrations also indicate dates upon which samples were collected and incubated for primary production estimates.

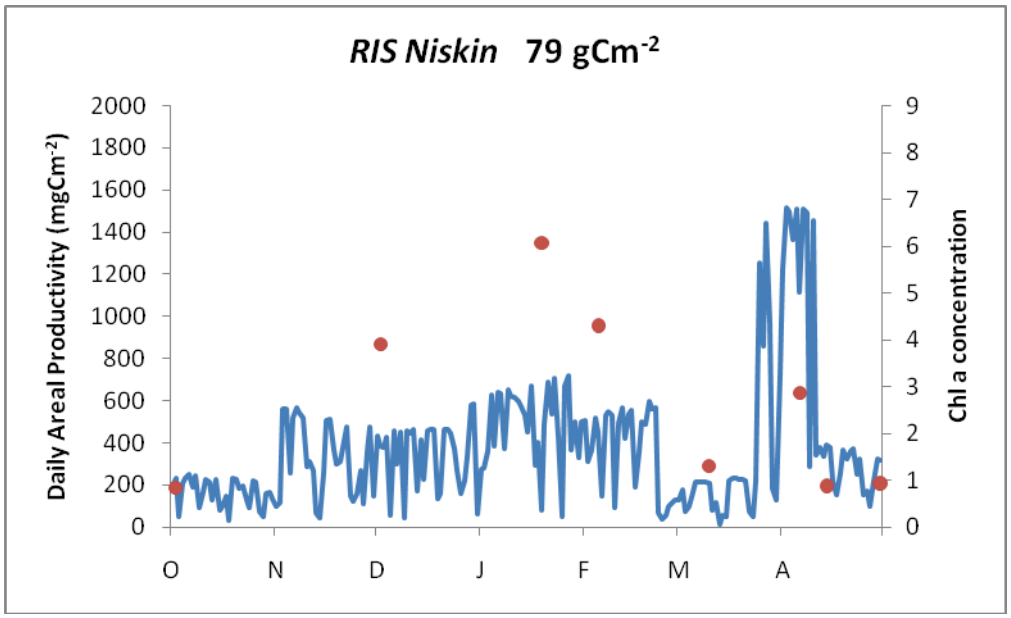

Fig 9. Daily Areal Production (blue lines) estimates and measured chlorophyll a concentrations (red dots) from RIS utiizing niskin sampling method. The dates corresponding to the chl a concentrations also indicate dates upon which samples were collected and incubated for primary production estimates.

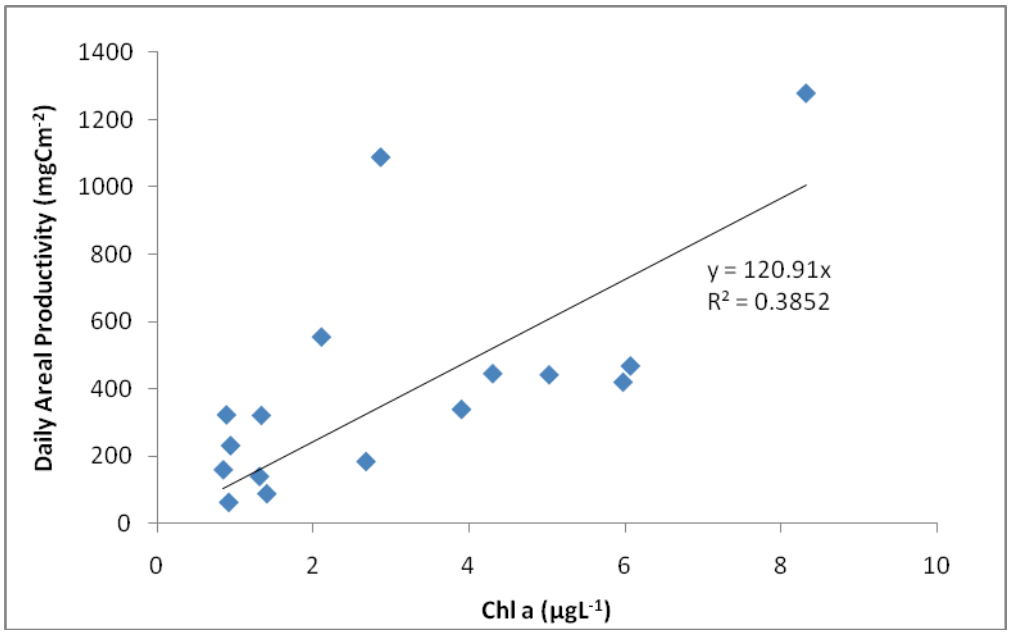

Fig 10. Production vs. Chlorophyll a concentration for samples from both sites collected with a Niskin water sampler.

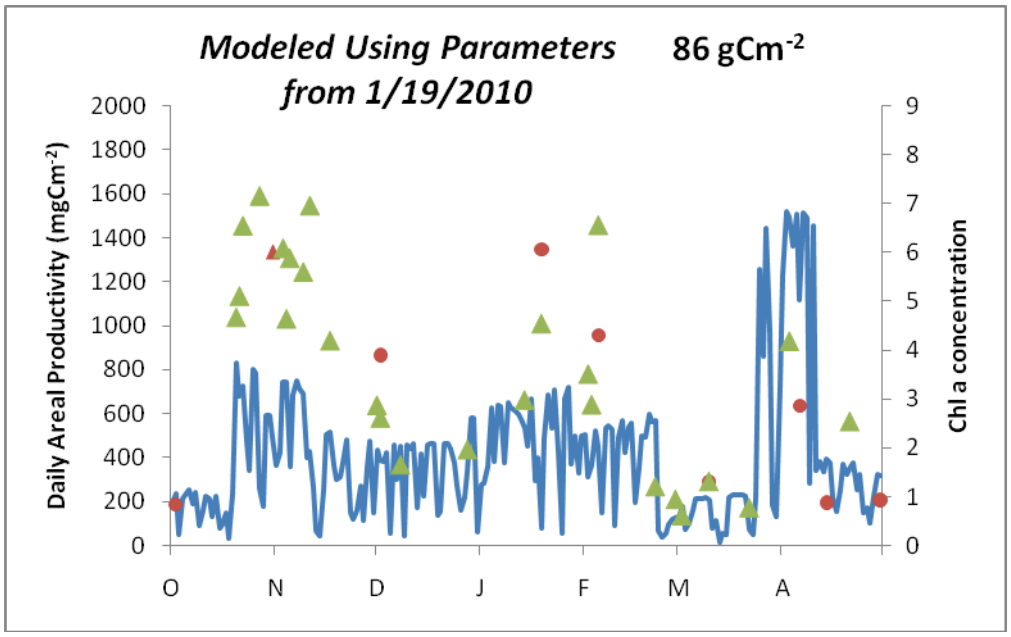

Fig 11. Daily Areal Production (blue lines) estimates for RIS using parameters from 1/19/2010 for the time period between 10/20/2009 – 11/11/2009 to attempt to correct for the missed phytoplankton bloom. Measured chlorophyll a concentrations on when production estimates were made are indicated by red dots and chlorophyll a concentrations on days that primary production incubations were not completed are indicated by green triangles.

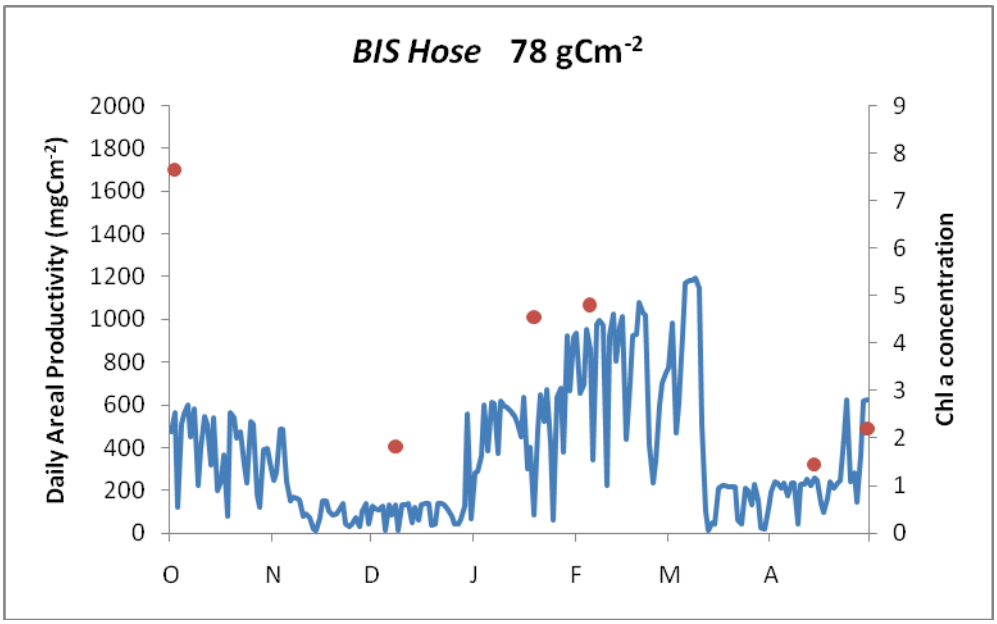

Fig 12. Daily Areal Production (blue lines) estimates and measured chlorophyll a concentrations (red dots) from BIS utiizing hose sampling method. The dates corresponding to the chl a concentrations also indicate dates upon which samples were collected and incubated for primary production estimates.

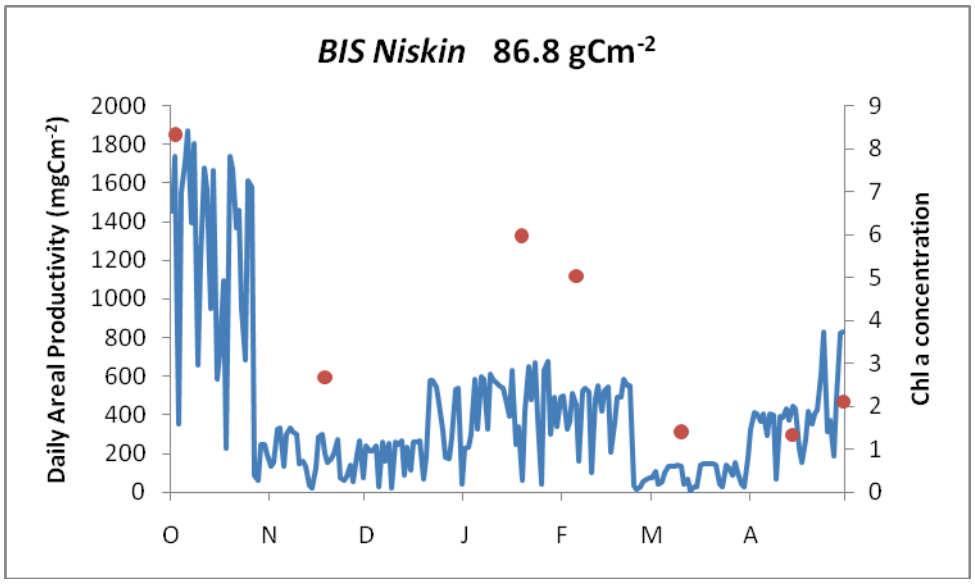

Fig 13. Daily Areal Production (blue lines) estimates and measured chlorophyll a concentrations (red dots) from BIS utiizing niskin sampling method. The dates corresponding to the chl a concentrations also indicate dates upon which samples were collected and incubated for primary production estimates.

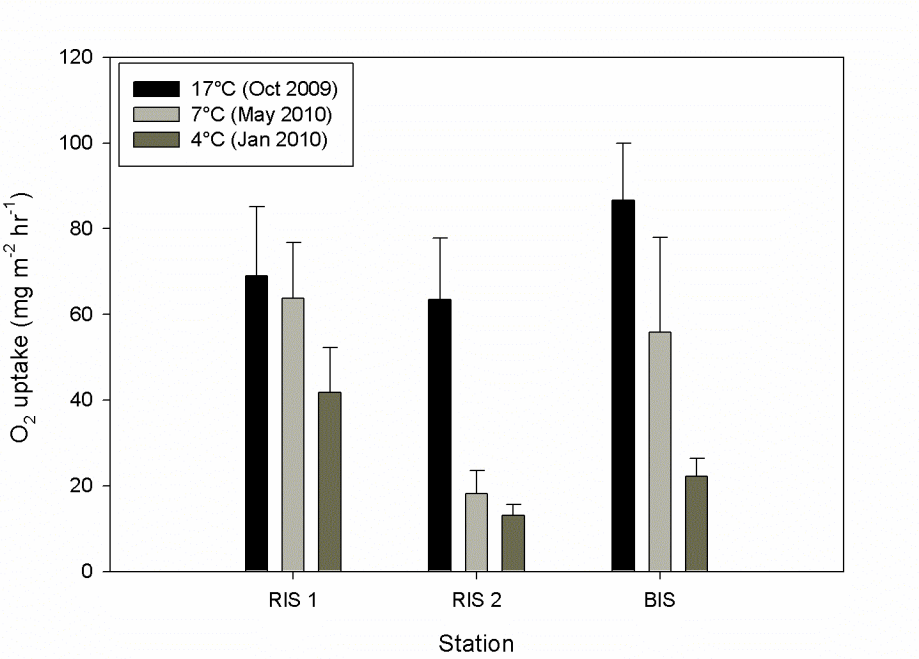

Fig 14. Mean sediment oxygen uptake measured through sediment core incubations at different temperatures (17°C, 7°C , and 4°C) for 2 stations in RIS and 1 station in BIS. Station RIS 2 is the easternmost RIS station, and RIS 1 is the western Rhode Island Sound station. Bars represent standard deviation.

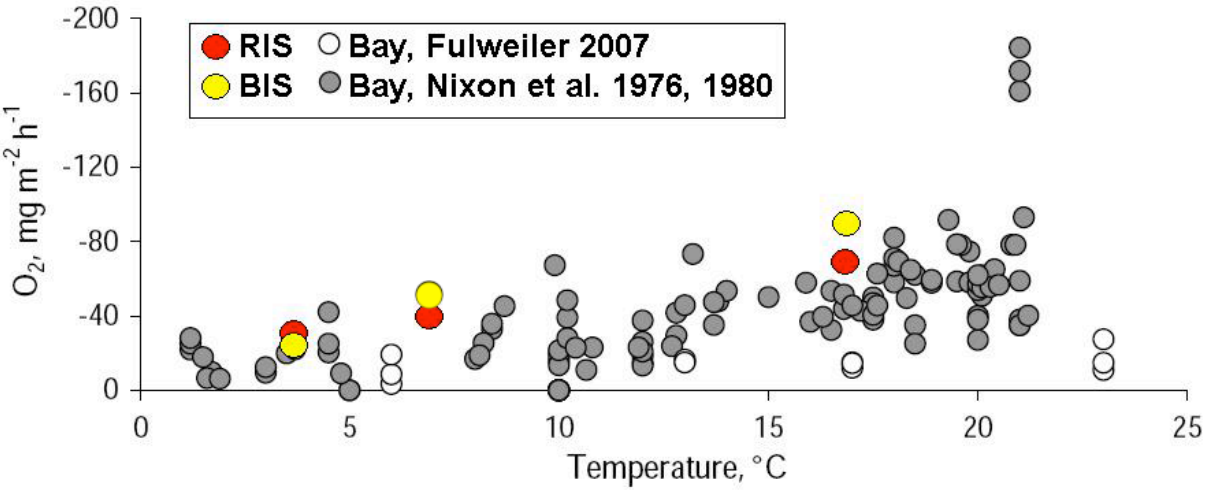

Fig 15. Sediment oxygen uptake for mid-Narragnsett Bay over an annual temperature cycle. Opaque circles are historical values (Nixon et al. 1976, 1980, and unpublished data), open circles are from more recent measurements taken in 2005-2006 (Fulweiler 2007). Red and yellow circles show mean sediment oxygen uptake for Rhode Island Sound (RIS) and Block Island Sound (BIS), respectively. Adapted from Fulweiler and Nixon (2009).