VARIATION IN MICROPLASTIC CONTENT OF MARSH SEDIMENT DUE TO THE ATLANTIC RIBBED MUSSEL *(GEUKENSIA DEMISSA)*, JAMAICA BAY, NY.

A Final Report of the Tibor T. Polgar Fellowship Program

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ABSTRACT

Microplastics are ubiquitous in the marine environment, yet their fate and impacts in estuaries are unknown. It is unclear if benthic suspension feeding can be a mechanism enhancing microplastic content in adjacent sediment. This study examined whether the Atlantic ribbed mussel (*Geukensia demissa*), would increase microplastic deposition in the salt marsh sediment of an urban estuary (Jamaica Bay, NY) via suspension feeding and biodeposition, or by altering hydrodynamics with their physical presence, or both. Three experimental treatments were created on Yellow Bar Marsh. Treatment one was the live mussel treatment to replicate mussels' suspension feeding and biodeposition. Treatment two was mussel shells to depict the mussels' physical presence, and treatment three was the control with no mussels. After approximately four weeks sediment samples were collected from each plot and a method was created to quickly and inexpensively extract microplastics with density separation and quantify microplastics with Nile Red staining. Live mussel treatments had significantly more microplastics (17% higher) than the empty shell treatment and the control treatment (19.22% higher). A 25.80% decrease in plastics was observed from August to September, suggesting fluctuations of microplastic content over time. These results indicate that the presence of active suspension-feeding ribbed mussels enhances microplastic content in Jamaica Bay marsh sediments. Thus, salt marshes with significant ribbed mussel populations may be a sink for microplastics within urban estuaries.

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INTRODUCTION

The mass production and use of plastic has substantially changed the modern world. Plastics are inexpensive, versatile, and durable and contribute to nearly every aspect of modern society. Worldwide production has grown considerably from 1.7 million tons in 1950 to 322 million tons in 2015 (PlasticsEurope 2016). Unfortunately, the qualities that make plastic so useful are also what make it so problematic. Due to the high durability and resistance to degradation, plastic is estimated to persist in the environment for hundreds of years and pose harm to the marine environment (Wang et al. 2016).

The National Oceanic and Atmospheric Administration (NOAA 2014) has defined microplastics (MP) as plastic particles <5 mm in size. MPs are a global environmental issue due to their ubiquity in the oceans, bioavailability, and ability to adsorb toxic chemicals (Shim et al. 2017). When initially introduced, MPs enter the marine environment as existing small particles (primary source) or degraded pieces of large plastic debris (secondary source). Primary sources of MP consist of microbeads found in hand and facial cleansers, cosmetic preparations, and pre-production plastic pellets (nurdles). Secondary sources are derived from fragmentation of larger plastics as a result of physical and chemical effects (Wang et al. 2016).

There is a large mass of MPs in the world's oceans. The 5 Gyres Institute used an oceanographic model of floating debris dispersal to estimate that a minimum of 5.25 trillion plastic particles weighing around 269,000 tons are in the world's oceans (Eriksen et al. 2014). Among global marine plastic debris, 92.4% of the items are MPs (Eriksen et al. 2014). The small size of MPs allows them to pass through sewage treatment plants, or to be

discharged from combined sewer outfalls (CSOs) unfiltered and to be potentially ingested by marine organisms, leading to bioaccumulation in the food web.

MPs float and transport long distances and are found everywhere from the near shore to open ocean, and from the sea surface to bottom (Shim et al. 2017). Plastics are comprised of different polymers and, depending on the composition, density, and shape, they can be buoyant, neutrally buoyant or sink. Although they are typically initially buoyant and float on the surface of the water, the density of the particles can increase with residence time in the water and enter the water column as neutrally drifting or slowly sinking particles (Wang et al. 2016). Some particles can sink further and become incorporated into the sediments **(**Lobelle and Cunliffe 2011). Estimates for the longevity of plastics in the environment are variable and depend on the physical and chemical properties of the polymer, as well as their physical location within the water column. For example, plastic degradation is slower in deep water and sediments with low oxygen concentrations and absence of light (Barnes et al. 2009).

Due to their small size, MPs are bioavailable to organisms throughout the food web (Cole et al. 2011). Ingested MPs have been reported in various organisms including filter feeders, under both controlled laboratory settings and in the wild (Shim et al. 2017). Vandermeersch et al. 2015 demonstrated that Mediterranean mussels (*Mytilus galloprovincialis)* ingest MPs in the field and in controlled lab conditions.

Suspension-feeding marine organisms initiate sedimentation of fine suspended matter. During suspension feeding, captured particles may be sorted prior to ingestion, and less nutritious particles can be rejected as pseudofeces (Newell 2004). This rejected material is transferred to the sediment along with feces as biodeposits (Bilkovic et al.

2017). Furthermore, ingested MPs can be packaged in feces and deposited on the sediment surface. In a controlled experiment, Khan and Presant (2018) demonstrated that 100% of Atlantic ribbed mussels (*Guekensia demissa*) used in their study ingested microplastics (Polystyrene spheres). Microplastics were found throughout digestive tracts of all specimens and feces, and pseudofeces from all experimental mussels contained MPs. Buoyant plastics become negatively buoyant when packaged in feces/pseudofeces, which suggests that these biodeposits are a source of MPs to the benthos (Khan and Prezant 2018). Thus, suspension feeding and biodeposition may represent an important mechanism for transferring MPs from the water column to the benthos.

Jamaica Bay is located at the southwestern end of Long Island, New York (Figure 1). The bay is protected by a barrier beach and it connects with lower New York Harbor and the Atlantic Ocean through the Rockaway Inlet at its western end. Jamaica Bay contains diverse ecologically important habitats including salt marshes, fresh and brackish water ponds, upland fields and woods, and islands.

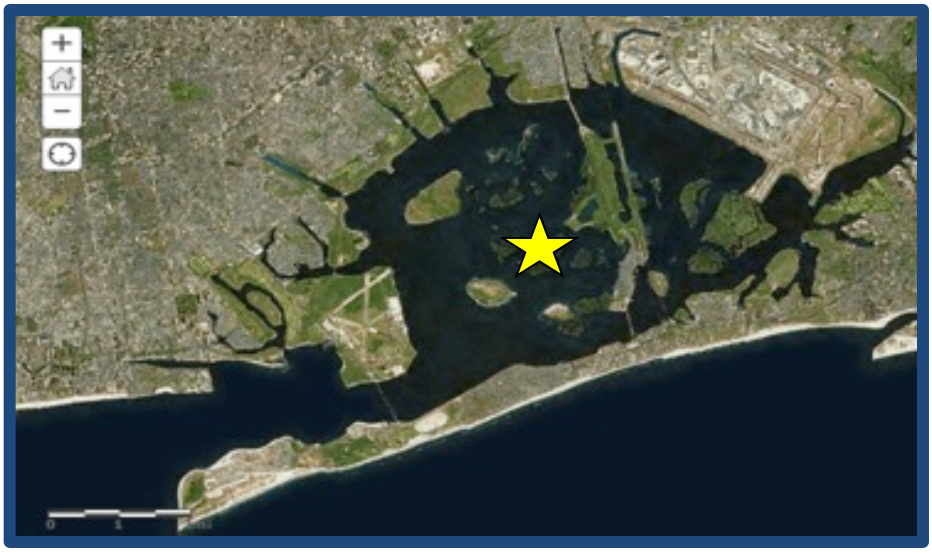

Figure 1. **Map of Jamaica Bay, with location of the study plots on Yellow Bar Marsh represented by the yellow star.**

Jamaica Bay is one of the largest areas of open space in New York City. The Bay is located within a densely populated region and has experienced significant anthropogenic alterations, including changes to the pattern of freshwater discharge. Freshwater input is largely derived from wastewater treatment plant (WWTP) effluent, CSOs, and storm drains, rather than natural streams (NYC DEP 2007).

A combined sewer system (CSS) collects rainwater runoff, domestic sewage, and industrial wastewater into one pipe. Under normal conditions, it transports all of the wastewater it collects to a wastewater treatment plant for treatment and then discharges it to a water body. The volume of wastewater can sometimes exceed the capacity of the CSS or treatment plant, such as during heavy rainfall events or snowmelt. When this occurs, untreated stormwater and wastewater discharges directly into a water body via CSOs (EPA 2016). CSOs are therefore often a source of pollution to receiving bodies of water. Several studies have determined that wastewater treatment plants and CSOs are a source of MP in marine environments. Estahbanati and Fahrenfeld (2016) sampled the Raritan River, in New Jersey, which receives effluent from more than 10 municipal WWTPs and concluded that the WWTPs are a contributing source of MP contamination. Carr et al. (2016) also found that effluent discharges from wastewater treatment facilities contribute to MP loads in oceans and surface water environments. Microbeads are not fully removed from the water phase during wastewater treatment and are therefore present in treated effluent and in receiving waters located downstream from WWTP outfalls (Wu et al. 2017).

There have been efforts to reduce sources of MP pollution. The Microbead Free Waters Act (2015) banned the sale of personal care products containing plastic microbeads, in the U.S., effective July 1, 2017. Other nations including Canada, Australia, and several

European countries are encouraging phase-outs or bans of plastic microbeads. More countries are likely to adopt similar a ban, thereby eliminating a major source of MPs (Wu et al. 2017). Monitoring of MPs in various abiotic and biotic environmental matrices can provide basic scientific information to determine their abundance, hot spots of concentration, historical trends, and exposure of organisms (Shim et al. 2017).

Atlantic ribbed mussels are an ecologically important species in Jamaica Bay. Ribbed mussels live in tidal marshes partly embedded in marsh sediment, attached to marsh grass roots, or along rock and shell beds. They are suspension- feeders and remove large amounts of particulate organic material such as algae, detritus, and bacteria from overlying waters through filtration and ingestion (Bilkovic et al. 2017). Suspension feeding processes a large amount of water, which maximizes exposure to harmful material within the water column, such as chemical pollutants and MPs (Vandermeersch et al. 2015).

Ribbed mussels are a prey species for blue crabs (*Callinectes sapidus*), mud crab, *(Panopeus herbstii*), willet (*Catoptrophorus semipalmatus*) and dunlin (*Calidris alpine)* thus providing a pathway for MPs to higher trophic levels (Nestlerode 2009). The beds assembled by mussels are a biogenic habitat that hosts a diverse population of immobile and mobile organisms (Sepúlveda et al. 2016).

Mussels may also be exposed to MPs in their food source. As filter feeders, mussels ingest phytoplankton, zooplankton, and algae. MPs have been observed inside plankton and zooplankton (Cole et al. 2013). Zooplankton are common in nearly all marine/estuarine ecosystems and can ingest MPs. The capacity for uptake varies between species, life-stage, and MP size. Lower-trophic level organisms are susceptible to ingesting MPs since they are indiscriminate feeders with limited ability to differentiate between

plastic particles and food (Cole et al. 2013). MPs can also accumulate on the external surface of dead zooplankton (Cole et al. 2013). Lower-trophic organisms such as zooplankton may facilitate the trophic-transfer of these contaminants up the food chain, with the potential for bioaccumulation and therefore adverse health consequences in higher trophic organisms (Cole et al. 2013). In a lab experiment by Farrell and Nelson (2013) results showed that, although a small amount, microplastic particles were transferred from mussels (*Mytilus edulis*) to crabs (*Carcinus maenas)*. This study demonstrated that trophic transfer occurs between mussels and crabs and that microplastic can translocate within the tissues (stomach, hepatopancreas, ovaries, and gills) of the crab (Farrell and Nelson 2013). MPs may also present a mechanical hazard to small organisms. Once MP is ingested, these fragments might block feeding appendages or hinder the passage of food through the intestines in small organisms, which causes pseudo-satiation and reduced food intake (Cole et al. 2013).

MPs are an emerging topic of research, and to date, no standard method has been developed for identifying and quantifying them in field samples. Although Fourier-transform infrared spectroscopy (FTIR) and Ramen spectroscopy can identify MP, they are expensive and time-consuming techniques (Shim et al. 2017). Nile Red, a fluorescent dye, is a useful dye for staining highly hydrophobic MPs. It has been used to stain neutral lipids in biological samples and synthetic polymers in polymer chemistry. Nile Red specifically binds to neutral lipids and is strongly fluorescent only in the presence of a hydrophobic environment (Shim et al. 2017). Nile Red staining methods were shown to be highly effective for identifying <100 mm particles (Shim et al. 2017).

The present study aimed to determine if the feeding behavior of Atlantic ribbed mussels increased the content of MPs in marsh sediments while using a quick processing method such as the Nile Red Dye technique. The hypothesis was that sediments associated with dense aggregations of live mussels would have higher MP content than sediment associated with mussel shells, or no mussels, and that the content may fluctuate temporally. The objective of the experiment was to determine if MP content in marsh sediment is increased by the physical presence and hydrodynamics of mussel shells alone, by the mussel's filter feeding behavior or both. Atlantic ribbed mussels are abundant in Jamaica Bay marshes and can reach densities of 10,000 per m^2 (Franz 2001). Their abundance in the bay has the potential to significantly impact the transfer of MPs from the water column to the benthos, making salt marshes a potential sink for MPs in this estuary.

METHODS

Experimental Design

Three experimental treatments were established on Yellow Bar Marsh in Jamaica Bay (Fig. 2). Treatment one was the live mussel treatment, which was intended to mimic a typical marsh bed where the mussels filter water and form biodeposits. Treatment two was empty mussel shells, which was intended to mimic the physical effect of mussels on the boundary layer hydrodynamics, which could trap MPs without the filter feeding. The empty mussel shells were collected and sealed back together with marine epoxy. Treatment three was the control with no mussels or empty shells. All three treatments contained saltmarsh cordgrass (*Spartina alterniflora*) and were replicated five times to create a total of 15 experimental plots (Fig. 3).

Figure 2: **Location of plot transects in Yellow Bar Marsh.**

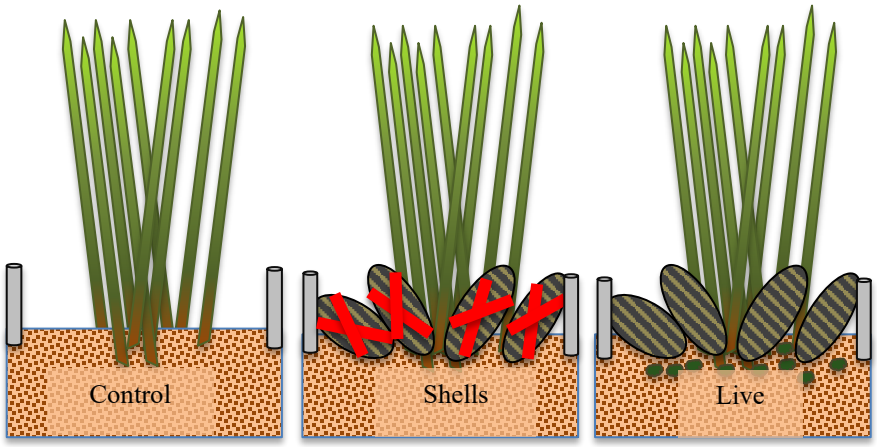

Figure 3: **Depiction of three treatments: Control, Shells and Live.**

On June 22nd, 2017 the locations of the fifteen 0.25m^2 experimental plots were established on the restored Yellow Bar Marsh in Jamaica Bay. The 15 plots were set up in two transects, and each plot was assigned a random number using a random number generator. For each plot, the corners were marked with a 1.27cm wide, 30.48 cm long

metal pipe. Each of the plots contained 18-22 stems of *S. alterniflora* for uniformity. On July 18, 2017, 400 live mussels or empty mussel shells were added to each plot for treatments 1 and 2 respectively, creating densities of 1,600 individuals per m^2. Most ribbed mussels tend to settle on aggregates of adult mussels around the stems of *S. alterniflora* and can reach densities of 2000–3000 in New England and 10,000 in Jamaica Bay, New York, per m^2 (Franz 2001); however, it was not possible to create this density in the experimental plots.

Sampling

The plots were left undisturbed for approximately four weeks before first sampling, then left another four weeks before final sampling. Each plot was sampled twice, once on August 14th, 2017 and once on September 18th, 2017, resulting in 30 samples, with 10 samples from each treatment.

The plots were divided into four quadrants and a different quadrant was sampled during each visit to avoid resampling the same area. A 9-cm diameter metal ring was placed at a depth of 1 cm into the sediments. The contents within the ring were extracted using a metal spatula and placed into a 118.3 ml Mason jar. This procedure was repeated twice within the selected quadrant at each plot to create a total sample area of 127.2 cm^2 per plot. When sampling the live mussel and empty shell plots, the individuals had to be lifted and sediments scraped from the shell. Five samples per treatment were collected each sampling period; however, one of the empty shell treatment samples from August 14th was lost during the lab processing.

Laboratory Methods

The protocol for separating and quantifying MPs in the sediment samples was adapted from the methods in Maes et al. (2017), Masura et al. (2015), and Tamminga et al. (2017). The samples were dried in their Mason jars overnight at 80°C while loosely covered with aluminum foil. Once dried, the samples were weighed and transferred to a 500 ml beaker for wet peroxide oxidation (WPO) with the Fenton reaction in order to digest and eliminate organic material (Masura et al. 2015). Equal parts (15ml) of aqueous iron 0.05 M FE(II) and hydrogen peroxide were added to the beakers containing the sediment sample and 10 ml deionized water. The mixture was allowed to rest on the lab bench at room temperature for five minutes prior to heating. The beaker was then covered with a watch glass and heated to 75°C on a hotplate while stirring for 35-45 minutes. If natural organic material was visible, another 15 mL of 30% hydrogen peroxide was added. This was repeated until no natural organic material was visible. Next, after being allowed to cool, the samples underwent density separation using a zinc chloride solution (Maes et al. 2017) prepared to a density of 1.38 mg/l. One hundred ml of aqueous zinc chloride was added to the sample in a 500 ml beaker, stirred vigorously with a spatula for 10 min, and allowed to sit for 15 minutes.

After 15 minutes, the liquid at the top was decanted into a glass funnel with connected tubing and a pinch clamp. Deionized (DI) water was used to rinse the sides of the beaker to ensure all the particles, including MPs, were washed into the funnel. The funnel top was covered with aluminum foil, and the sediment was allowed to settle overnight. Then, the settled solids were first drained through the tubing using the pinch clamp and discarded. The remaining liquid was collected in a clean 500 ml beaker. To treat

the sample with Nile Red dye, 50µL of Nile red stock was added per 5ml (0.25 ml to 25 ml) of water. The samples were then put on a shaker table for 60 min at 100 rpm. The Nile Red stock was prepared to a concentration of 1 mg/ml in acetone (Tamminga et al. 2017). After the sample had incubated for 60 min, it was vacuum filtered onto a 0.7µm GF/F filter pad. The filter was placed in a petri dish with a cover to dry. The filters were viewed under 10x magnification with an Olympus BX51 microscope under blue light fluorescence. The Nile Red stained particles fluoresce when exposed to blue light with a wavelength of 410-450 nm (Maes et al. 2017) (Fig. 4).

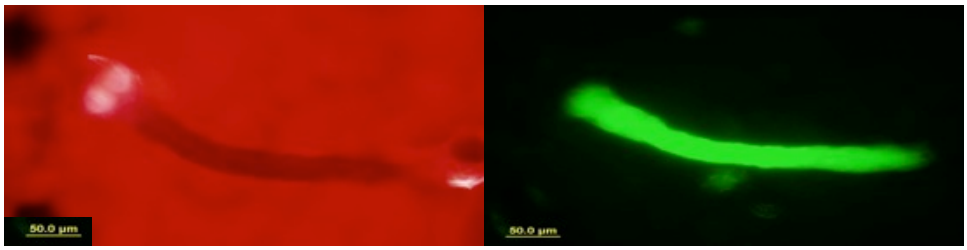

Figure 4. **The left image depicts a MP fiber under bright light. The image on the right is the same MP fiber under blue light. The MP fiber was found in a Live Mussel plot sample from August 2017.**

Counting every piece of plastic on the filters would be prohibitively time consuming. Therefore 100 random fields of view (FOVs) through the microscope were counted on each filter pad, to determine an average amount of particles per FOV. This average was then extrapolated over the entire area of the filter pad. At 10x magnification, the FOV is a 2.2 mm diameter circle with an area of 3.80 mm^2. The effective area of the 47 mm Whatman GF/F filter pads is a 42 mm circle with an area of area of 1385 mm^2. Thus, each FOV is 1/365 of the entire filter pad area. To extrapolate for the entire filter pad, 365 multiplied the average MPs per FOV. The total number of MPs on the filter pad was

divided by the total plot sample (127.2 cm^2) and reported as MP/cm^2 ($\#MP/cm^2$) of sediment.

Statistical Methods

The data were analyzed in RStudio (RStudio Team 2016). A two-way-analysis of variance (ANOVA in Stats Package; Chambers et al. 1992) was used to test for significant differences among the treatments and over time. A post hoc Tukey HSD test (Stats Package -Douglas Bates) was used for pairwise comparisons among the treatments.

RESULTS

Average MP content per square centimeter of sediment surface was highest in the live treatments (Fig. 5). It was expected that sediment MPs would increase from August to September due to accumulation. However, there was 25.80% less MPs in the September samples in comparison to the August samples (Fig. 6); this loss could be due to changes in the net deposition. When all samples from August and September were combined for each treatment, the average MP concentration in the live treatments was 17% higher than the empty shell treatment and 19.22% higher than the control treatment (no shells). In August the average MP concentration in the live treatments was 16.81% higher than the shells, and 22.40% higher than the control. In September the average MP concentration in live plot samples was 14.47% higher than the shells, and 13.34% higher than the control.

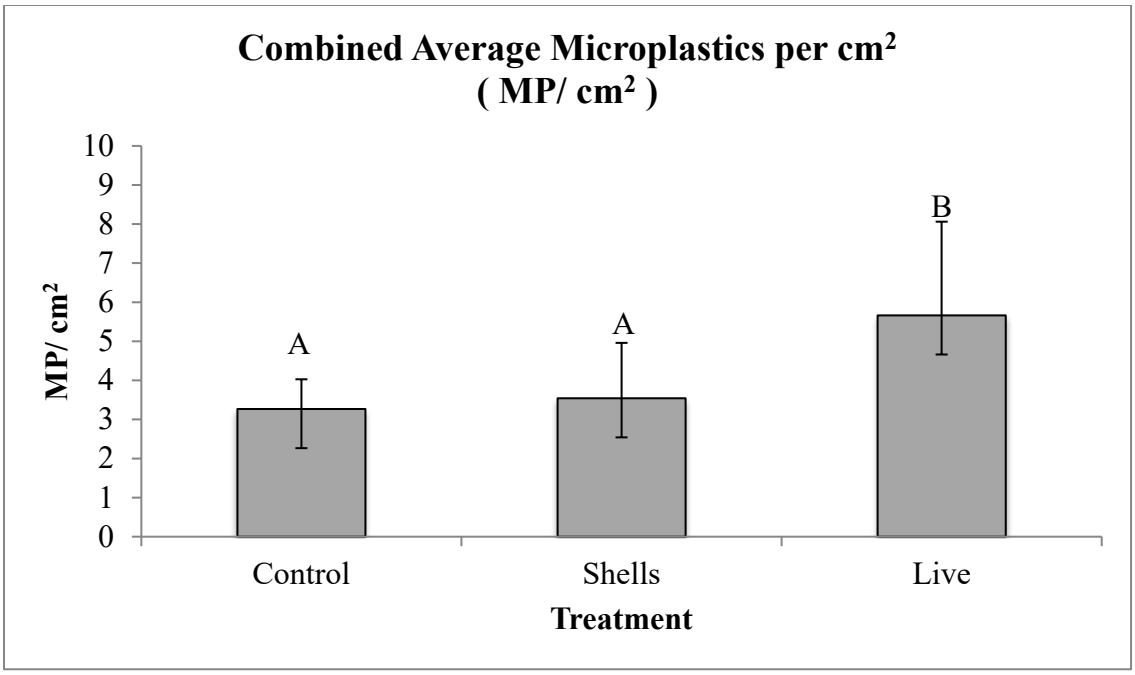

Figure 5. **Combined average MPs per cm² of sediment collected in August and September 2017.**

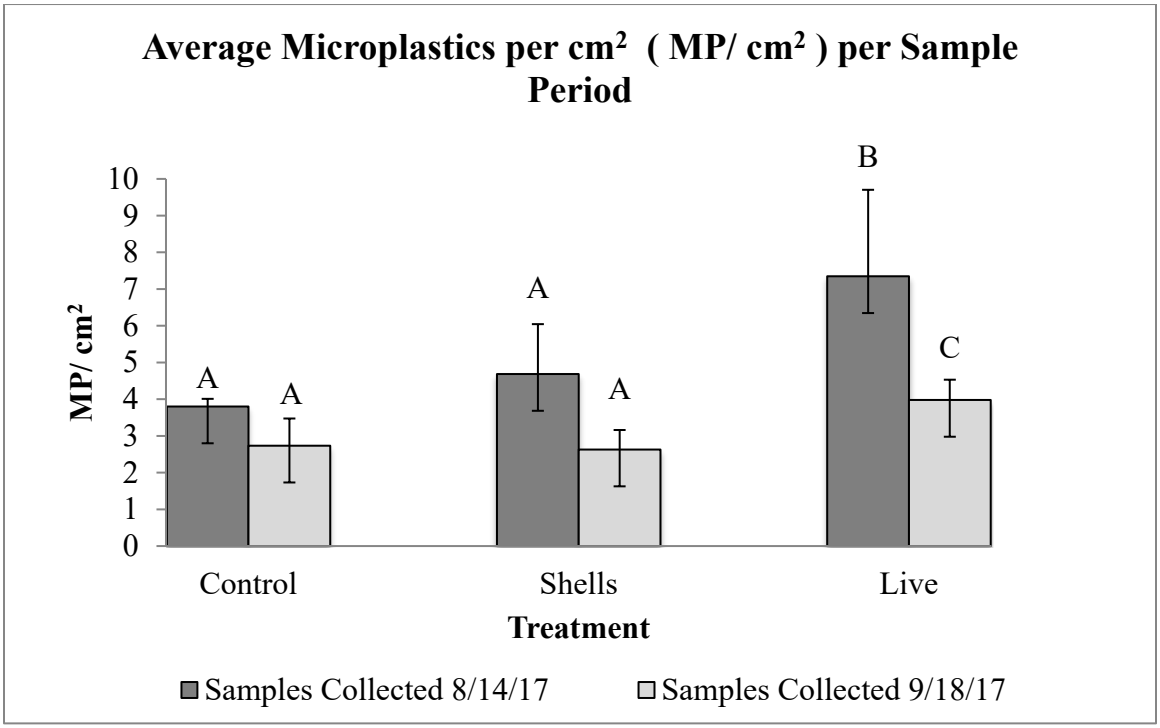

Figure 6. **Average MPs per cm² of sediment collected in August and September 2017.**

Statistical Results

The results from the two-way analysis of variance (ANOVA) and post hoc Tukey HSD test are presented in Table 1. There was a significant difference among treatments (ANOVA: $p<0.001$) and over time (ANOVA: $p<0.001$). There was no significant interaction between treatment and time. A post hoc Tukey HSD test indicated a significant difference between the live mussel plots and empty shell plots ($p<0.05$), and live mussel plots and control ($p <.001$), but no significant difference was noted between control plots and empty shell plots ($p >0.05$). The decrease in MP content was significant over time (ANOVA: $p<0.001$). A significant difference was observed in the loss of MPs in live plots over time ($p <0.05$) but not in shell or control plots ($p >.05$)

Table 1. Results from the Two-Way-ANOVA and pairwise post hoc Tukey HSD tests of differences among treatments and over time. *$p < 0.5$; **$p < 0.01$

2 way ANOVA	***F***	***p***		**Tukey post hoc**	***p***
Among Treatments	12.012	<0.001 **		Control-Shells	>0.05
Between Months	24.016	<0.001 **		Control- Live	<0.001 **
Treatments: Month	2.354	0.11		Shells - Live	<0.05*
				September- August	<0.001 **
				Live Aug - Live Sept	<0.05*
				Control Aug - Control Sept	>0.05
				Shells Aug - Shells Sept	>0.05

DISCUSSION

The results support the hypothesis that sediments associated with dense aggregations of live mussels will have higher concentrations of MPs in comparison to sediment associated with just empty shells (no live mussels), or no mussels at all. There were significantly more MPs in the live mussel plots as compared to the control and shell plots, indicating that sediment MP abundance was affected by the presence of the live ribbed mussels. A plausible explanation for this result is that the feeding behavior of the mussels, and not the sheltering effect and hydrodynamics of their shells in the boundary layer, resulted in increased MP deposition. These results corroborate Khan and Prezant's (2018) study, which determined that plastics become negatively buoyant when packaged in feces/pseudofeces, suggesting that the biodeposits are a source of MPs to the benthos.

MP content decreased overall by 25.80% between the September and the August samples. The net content of MP may change over time. Marsh surfaces accrete vertically through deposition of tidally delivered sediment, growth of belowground plant parts, and accumulation of litter from aboveground plant growth. Marshes also lose elevation as a result of decomposition, erosion, and compaction (Anisfeld and Hill 2012). Temporal fluctuations in MP content could result from sediment and particle re-suspension affecting the net MP content. Changes in wind and temperature are two potential factors that could have caused the decrease. MPs that were deposited could be dispersed by wind and waves with re-suspension of particles. Local temperature changes could have affected the mussels feeding behavior over time in the plots. It could be that although the feeding behavior of mussels tends to deposit MPs onto the sediment surface, wind or waves may disperse the

sediment and associated MPs. Since wind, tides and currents can transport and re-suspend sediment (Imran 2008), it is likely that they would transport MP particles as well.

Weather conditions following the first sampling event, such as high wind events and storms, could have altered the plots and dispersed the MPs. To evaluate this potential mechanism, average wind speeds recorded at the John F. Kennedy Airport weather station, which is located 7 km northeast of the plots, were obtained from National Center for Environmental Information (NCEI) and analyzed (NCEI 2017). Although no extreme variances in the wind speeds were observed during sample periods, the wind speeds following the second sample had more days with faster winds than after the first sample date (Fig.7).

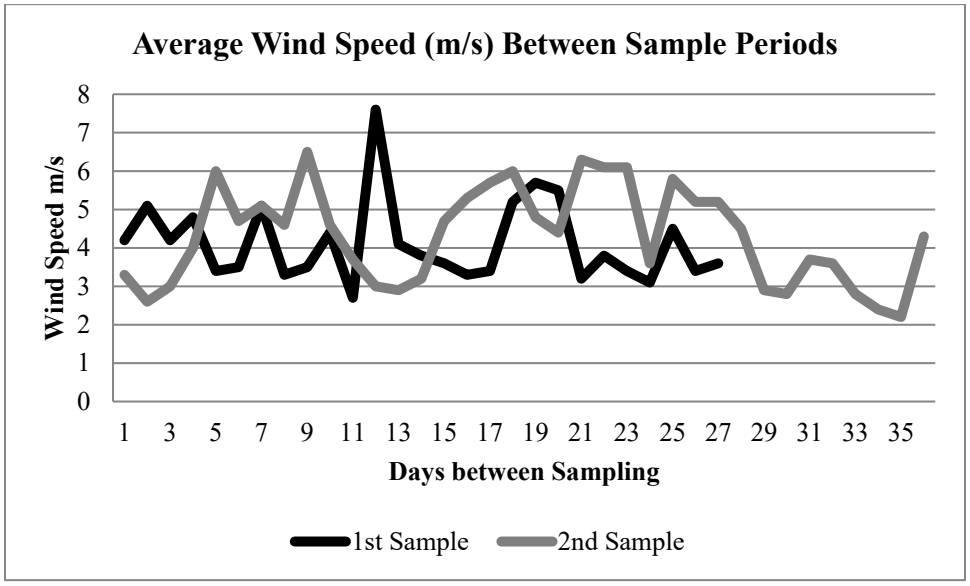

Figure 7. Average wind (m/s) preceding plot set up and first sample date (NCEI 2017, JFK Airport, NY).

Wind direction could also significantly impact the plots. The dominant wind direction over each sampling period was a South wind (Fig. 8). With the plots located along the marsh edge a wind from the south could affect them. Both wind speed and direction

were similar between both sampling periods. Thus, there was no apparent strong wind event that could account for dispersal of MPs between sampling periods. Another factor that could have disturbed the plots is boat traffic adjacent to the marsh creating local waves.

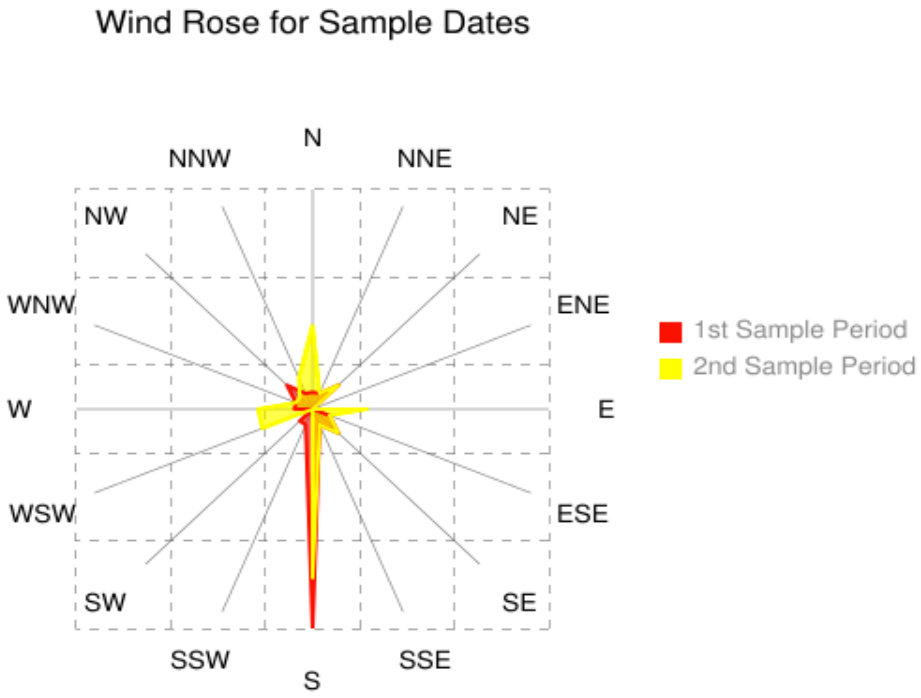

Figure 8. **Frequency of days of wind direction, with dominating southern winds (created on Enviroware.com; NCEI 2017, JFK Airport, NY).**

Filtration rates of suspension feeders depend on several factors, including water temperature (Bilkovic et al. 2017). Temperature is an important factor for the metabolism and rate of feeding of marine bivalves (Huang and Newell 2002). The rates of aquatic respiration and physiological functions for Atlantic ribbed mussels vary seasonally, primarily due to temperature changes (Huang and Newell 2002). Seasonal changes in

temperature are known to directly affect the clearance rates of suspension-feeding bivalves, and lower temperatures suppress feeding activity (Kreeger and Newell 2001). It is reasonable then to expect that a decrease in water temperature between sampling dates affected mussel feeding behavior. A water logger deployed on site recorded a combination of air and water temperature (°C) every 15 minutes. The average temperature during the first sampling period of July installation until the August sampling was 24.83°C, and the average temperature during the second was 21.88°C. A pattern of decreasing and lower temperatures was observed during the second sample period (Figure 9). The first period often had temperatures of 24-28°C, while the second period ranged 18-23°C. This decrease and difference in temperature could have negatively affected mussel feeding rates thus filtering less MPs out of the water.

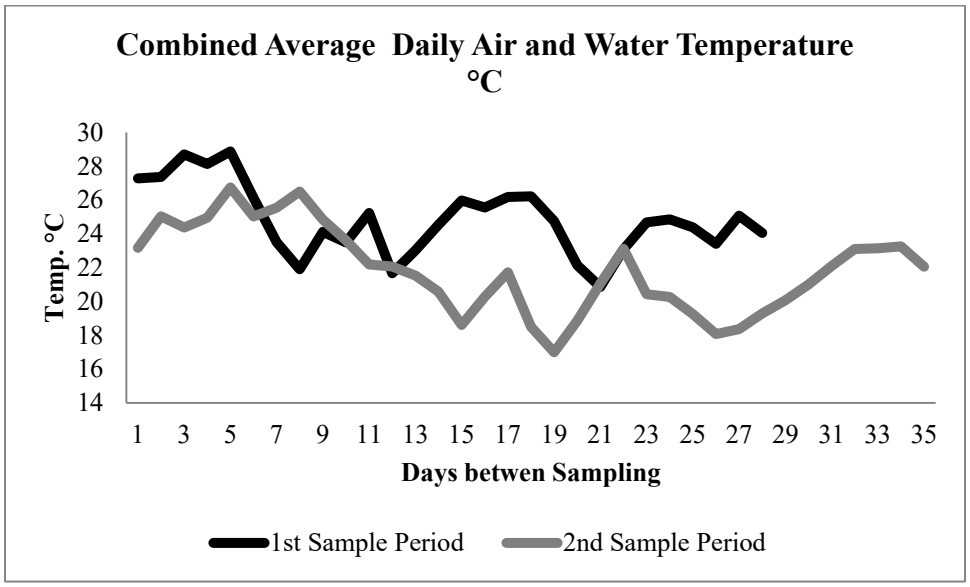

Figure 9. Temperature variations at the study site preceding plot set up and first sample date, with lower temperatures during the 2nd sample period.

From these results, an estimated minimum net microplastic deposition rate can be

estimated with a key assumption that all plots started with the same amount of MPs. By taking the average in all combined live plots of 5.66 MP/cm^2 and subtracting the control (3.27), extrapolating that to the entire 0.25m^2, and dividing that number by the 400 mussels, an average amount of MP deposition contributed by one mussel in a 0.25m^2 area was determined over the sample period. From there, an estimated number of MPs contributed by one mussel per day was determined by dividing by the days in the sample period. This equates to an average of 0.43 MPs contributed by one mussel per day. The experimental plots were less dense (1,600 per m^2) than typical mussel aggregation density in Jamaica Bay due limitations of experimental design. With New England mussel beds reaching densities of 3,000 mussels per m^2, which could equal 1,290 MP net particles deposited per m^2 per day. Jamaica Bay mussel beds can support densities up to 10,000 per m^2, which could equal 4,300 MPs deposited per m^2 per day.

Khan and Prezant (2018) estimated MP deposition ranging between 4,500 to 35,000 pieces/m^2 in NJ Atlantic ribbed mussel beds and noted considerable variation in abundance and particle size across sampling sites, sampling depth, transect locations. Although, they only sampled sites with mussels, they found that sites with higher mussel densities had greater sediment MP concentrations (Khan and Prezant 2018).

Overall, MP content is affected by the presence of the ribbed mussel, a suspension feeding organism. Ribbed mussels appear to filter MPs from the water and egest them with their biodeposits on the sediment surface. There are potential management implications for Jamaica Bay, based on these results. The presence of dense ribbed mussels appears to be a mechanism for increasing MP content in salt marshes by suspension feeding and local

hydrodynamics. Implementing mussel beds to work as bio-filters near sources of MPs, such as CSOs could potentially reduce the amount entering the estuary.

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