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Microplastics in Long Island Marine Estuaries

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Abstract of the Thesis

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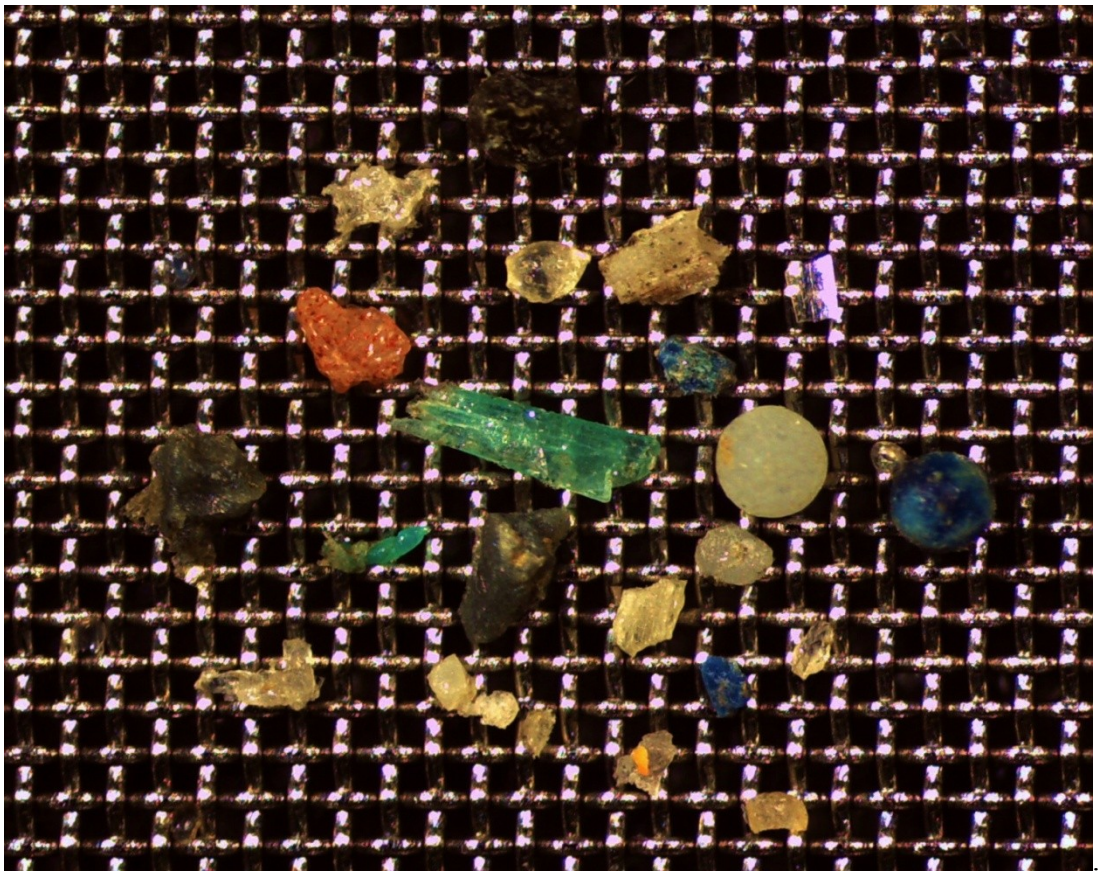
This study investigated the prevalence of microplastics in Shinnecock Bay and Jamaica Bay on Long Island in New York. Surface water samples were collected with a 200 μm plankton tow net, filtered onto metal mesh screens, and examined under a dissecting microscope. A total of 517 microplastic particles were found in samples from Shinnecock Bay (0-0.58 particles m^{-3}), and 1,005 particles were found in Jamaica Bay samples (0-3.93 particles m^{-3}). Particles were visually sorted by size, color, and shape. Statistical analyses were performed to analyze microplastic particle diversity, evenness, and any spatial or temporal variation. Attempts to determine source inputs were inconclusive, but it is likely that Jamaica Bay has point sources of microplastics in the forms of WWTP outfalls and CSOs, and Shinnecock Bay has nonpoint sources from land areas surrounding the bay.

Frontispiece



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List of Abbreviations

ANOVA - Analysis of Variance

BPA - Bisphenol A

CSO - Combined Sewer Overflows

DDD - dichlorodiphenyldichloroethane

DDE - Dichlorodiphenyldichloroethylene

DDT - Dichlorodiphenyltrichloroethane

HDPE - High-Density Polyethylene

K-S - Kolmogorov-Smirnov

LDPE - Low-Density Polyethylene

NP - Nonylphenol

PAH - Polycyclic Aromatic Hydrocarbons

PBDE - Polybrominated Diphenyl Ether

PCB - Polychlorinated Biphenyl

PE - Polyethylene

PET - Polyethylene Terephthalate

POP - Persistent Organic Pollutant

PP - Polypropylene

PS - Polystyrene

PVC - Polyvinyl Chloride

UV - Ultraviolet

WWTP - Wastewater Treatment Plant

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Chapter 1 Microplastics in Marine and Estuarine Environments: an overview

Introduction

Plastic debris has been entering the oceans for decades due to increased global production and use of plastics and poor waste management practices (Cole et al. 2011). In the North Atlantic and North Pacific Oceans, amounts of plastic debris have rapidly increased since the 1960's and 1970's (Moore 2008, Barnes et al. 2009), paralleling the five-fold increase in global plastics production from about 50 million tons in 1976 to about 280 million tons in 2011 (PlasticsEurope 2012). Now plastic debris has been found in every ocean and on shorelines of every continent (Browne et al. 2011). The majority of plastic is made of high- or low-density polyethylene (HDPE or LDPE), polypropylene (PP), polyvinyl chloride (PVC), polystyrene (PS), or polyethylene terephthalate (PET) (Andrady & Neal 2009). Plastics also contain numerous additives that are mixed in with the base polymer at time of production and include plasticizers, fillers, thermal stabilizers, fire retardants, UV stabilizers, colorants, and opacifiers.

Plastic in aquatic environments can negatively impact organisms in multiple ways, including entanglement, ingestion, toxicity, reduced foraging efficiency, and as a vector for invasive species (Barnes 2002, Derraik 2002, Moore 2008, Aloy et al. 2011, Choy & Drazen 2013). Green sea turtles (*Chelonia mydas*) in the Río de la Plata estuary have been found with plastic debris throughout their intestinal tracts (Gonzáles Carmen et al. 2014), and 25% of 192 longnose lancetfish (*Alepisaurus ferox*), a piscivore from the North Pacific, were found to contain plastic pieces in their stomachs (Jantz et al. 2013). Seabirds are particularly vulnerable to impacts from plastic, especially young birds who are accidentally fed plastic from their parents' foraging trips in the open ocean. Seabird chicks are often found starved to death with

stomachs full of plastic debris. There are many ongoing efforts to monitor plastics in the environment, both on beaches and in open waters, but these efforts can be costly, inefficient, and the methods used are not always standardized (Ryan et al. 2009).

In addition to large pieces of plastic debris in the oceans, there are also very small pieces called microplastics, defined as plastic particles and fragments 5 mm or less in size (Betts 2008). The presence of microplastics in the ocean was first discovered in the 1970's (Carpenter & Smith 1972), and awareness of them as an environmental issue has increased over subsequent decades not only within the scientific community, but also with the general public. Various types of legislation attempting to reduce the amount of plastics entering the marine environment have been put in place, such as bottle deposits to encourage recycling, restrictions on plastic bag use (Romer 2008), and MARPOL Annex V— an international law that prohibits vessels from dumping plastic waste at sea (Cole et al. 2011). Currently, lawmakers from different states, such as New York and California, are suggesting new legislations to ban microplastics from consumer products in order to reduce their damage to the environment, fueled by increased public concern for this novel source of pollution.

Sources and Characterization

There are two general classifications of microplastics, termed primary and secondary (Gouin et al. 2011). Primary forms of microplastic include scrubber particles, pre-production pellets that are used to make plastic products, and other particles that are less than 5 mm when they initially reach the ocean (Barnes et al. 2009, Fendall & Sewell 2009, Browne et al. 2011). As primary microplastics, polyethylene (PE) particles are referred to as nibs or nurdles, and PS spherules—the raw material for Styrofoam—are called suspension beads (Colton et al. 1974,

Wilber 1987). These particles are used to produce other plastic and Styrofoam consumer products and can enter the marine environment through terrestrial runoff, mishandling during shipping, or wastewater from plastic-production plants. PE nurdles tend to float on the surface while PS suspension beads are denser than water and sink, unless they contain air bubbles (Wilber 1987).

Residential wastewater effluent is a major source of primary microplastics and can contain synthetic fibers and micro-scrubber particles (Habib et al. 1998, Fendall & Sewell 2009, Browne et al. 2011). Synthetic fibers such as Nylon, Dacron, Orlon, and Spandex in washing machine wastewater can flow into ocean waters with effluent from sewage treatment plants (Habib et al. 1998, Browne et al. 2011). Some facial cleansers contain micro-scrubber particles for exfoliation that are made of PE and travel through wastewater into the marine environment (Gregory 1996, Fendall & Sewell 2009), where plastic fragments and fibers as small as 1 μm in diameter have been found (Frias et al. 2010). Due to their small size, many microplastics are not removed from wastewater at sewage treatment plants and thus make their way into the oceans (Moore 2008, Fendall & Sewell 2009). There appear to be no studies examining the fate of microplastics in septic systems that do not have direct flow into a water body, but it is likely that some of the particles may be trapped in septic tanks, while others may eventually seep out into water bodies with groundwater flow. Of even greater concern are the many parts of the world that have minimal or no sewage treatment at all before waste flows into rivers and ultimately, the oceans.

Secondary microplastic is formed by the fragmentation of macroplastic (Betts 2008). Plastic in the ocean does not biodegrade but photodegrades and weathers into smaller and smaller pieces (Andrady 2011). Marine organisms can also create microplastic fragments from

larger pieces in their environments by biting them or tearing off smaller pieces (Davidson 2012, Carson 2013). Photodegradation is caused by ultraviolet (UV) radiation at the surface of the ocean, where buoyant plastic is most commonly found. The breakdown by UV radiation is inhibited, however, by salinity and cold temperatures, and by marine organism fouling (Barnes et al. 2009, O'Brine & Thompson 2010). As a result, plastic can remain intact in the environment for hundreds to thousands of years, and even longer at depths where there is no UV light. Even if the addition of plastic to the ocean environment was halted, the number of plastic pieces would continue to increase because the plastic already present would keep photodegrading into smaller particles (Doyle et al. 2011).

Review of Global Sampling Studies

A number of global studies have examined beaches and subtidal areas to quantify microplastic abundance on shorelines. Browne et al. (2010) sampled beach sediments of the Tamar Estuary in the United Kingdom and found 952 pieces of plastic debris, 65% of which was microplastic smaller than 1 mm. High abundances of microplastics were also found in Belgian coastal marine sediments with freshwater rivers as potential sources (Claessens et al. 2011, Van Cauwenberghe, Claessens, et al. 2013). A remote Brazilian beach had both plastic fragments and pellets in the strandline, indicating long-range transport of which these particles are capable in the open ocean (Costa et al. 2010). Frias et al. (2010) detected persistent organic pollutants (POPs) ranging in concentration from 0.01 to 319.6 ng ¯g on microplastic particles found on two beaches in Portugal. McDermid & McMullen (2004) collected over 19,000 microplastic particles from beaches in the Hawaiian Islands, and Ng & Obbard (2006) identified and quantified low concentrations of microplastics on beaches in Singapore, the first study of

microplastics in an Asian marine environment. Vianello et al. (2013) sampled tidal sediments from the Lagoon of Venice in the Mediterranean Sea and found high abundances of microplastic particles, which were also correlated to fine sediment deposition and metal pollution index. Microplastics have even been found in deep-sea sediments at depths of 1100-5000 m (Van Cauwenberghe, Vanreusel, et al. 2013). Increased abundances of plastic fragments in beach sediments can alter the permeability and heat transfer of these sediments, potentially impacting organisms utilizing beach environments (Carson et al. 2011). These studies demonstrate the high prevalence as well as some of the negative effects of microplastics in sediments and coastlines.

In addition to sediment sampling, surface waters of the oceans are frequently sampled for microplastics. Though more sampling has been conducted historically in the Pacific Ocean, microplastics have been studied in the Atlantic Ocean as well. Carpenter and Smith (1972) sampled the surface waters of the Sargasso Sea with a 330 μm neuston net and found an average of 3,500 particles of plastic per square kilometer. Over a decade later, Wilber (1987) towed surface neuston nets across the northwestern Atlantic and Sargasso Sea and found more than double the concentrations of plastic found by Carpenter and Smith (1972) and Colton et al. (1974). The waters north of the Gulf Stream had about 700 pieces of plastic per square kilometer, believed to originate mostly from coastal processes rather than Gulf Stream and gyre circulation (Wilber 1987). A 22-year-long study by Law et al. (2010) found that 62% of 6,136 plankton tows conducted in the Northwestern Atlantic using a 335 μm net had plastic debris. Morét-Ferguson et al. (2010) analyzed a subset of these samples and found that density values were suggestive of PP and PE with increased densities from biofouling. They also found that densities of pelagic plastic particles varied from those of beach plastics, implying that residence time at sea can modify microplastic particles. The highest concentrations of microplastic found

by Law et al. (2010) were within the subtropical convergence in the surface velocity field that crosses the subtropical North Atlantic basin, due to strong currents such as the Gulf Stream rapidly moving particles offshore to the center of the subtropical gyre. In Portuguese coastal waters, 61% of zooplankton net tows contained microplastic particles (Frias et al. 2014).

Studies of plastics in waters around Long Island are few and outdated. Carpenter et al. (1972) found polystyrene spherules 0.5-2 mm in size in the waters of Long Island Sound and Block Island Sound. In a subsequent study, Austin and Stoops-Glas (1977) tracked the seasonal distribution and sources of nurdles in Block Island Sound. They found that the nurdles were most abundant in the late spring and early summer, most likely originating from plastic-manufacturing on rivers of Connecticut that drain into Long Island Sound. Colton et al. (1974) sampled coastal waters along the southern shore of Long Island and in Block Island Sound with 0.947 mm neuston nets and collected 50-100 grams of plastic per square kilometer. Most recently, synthetic fibers from sewage treatment plant effluent were found in the sediments of Huntington Harbor and Oyster Bay Harbor by Habib et al. (1998). To date, no previous studies of microplastic concentrations, characteristics, or distributions have been conducted in any of the Long Island south shore bays.

Microplastic-Organism Interactions

Just as with macroplastic, microplastics can be ingested or adsorbed by marine fauna and can impact marine food webs at multiple trophic levels. An organism that ingests macro- or microplastic can experience gut blockage, starvation, and ultimately mortality, as well as facilitate the mobility of non-nutritious plastic up the food web (Moore 2008, Wright et al. 2013). There have been many studies examining this phenomenon, most using feeding

experiments under laboratory conditions, and almost all of them conducted within the last decade. Bhattacharya et al. (2010) found that positively charged 20 nm PS particles could adsorb to *Chlorella* spp. and *Scenedesmus* spp. algal cells and inhibit their ability to photosynthesize. Similarly, two microalgal species, *Rhodomonas baltica* and *Tetraselmis chuii*, experienced growth inhibition and showed signs of oxidative stress from microplastics and the polycyclic aromatic hydrocarbon (PAH) pyrene (Oliveira et al. 2012). Thirteen zooplankton taxa, including four calanoid copepods, a cnidarian, a tunicate, a euphausid, decapod and bivalve larvae, and the dinoflagellate *Oxyrrhis marina*, were found to ingest microplastics by Cole et al. (2013). The copepods experienced decreased algal feeding rates in the presence of microplastics and adherence of the plastic beads to their external surfaces. Copepods, cladocerans, mysids, rotifers, polychaetes, and the ciliate *Tintinopsis lobiancoi* were shown to ingest fluorescent 10 µm PS spheres, which were still detectable after the zooplankton had been eaten by the mysid shrimp *Mysis relicta* (Setälä et al. 2014). The copepod *Tigriopus japonicus* experienced negative impacts on survival and fecundity across multiple generations from ingestion of PS microbeads (Lee et al. 2013).

The blue mussel *Mytilus edulis* has been examined extensively in microplastics studies, as it is commonly found in nearshore environments, is easy to raise and work with in laboratory settings, is a good species to monitor for environmental health, and some of the negative impacts on it from micro- and nano-particles are known (Canesi et al. 2012). Browne et al. (2008) fed blue mussels plastic particles 3 µm and 9.6 µm in size that were ingested within 12 hours; the particles were found in the mussel circulatory fluid after 3 days and remained in the mussels for as long as 48 days. Blue mussels have also ingested aggregates of 30 nm PS particles that reduced their feeding activity (Wegner et al. 2012). In one experiment, high-density PE beads

ranging > 0-80 μm in size were ingested by blue mussels, causing formation of granulocytomas in the connective tissue of the digestive gland and an overall reduction in health (von Moos et al. 2012). Farrell and Nelson (2013) fed fluorescent 0.5 μm microspheres to blue mussels, then fed the mussels to the green crab *Carcinus maenas*. They were able to detect the fluorescent particles in the stomach, haemolymph, and tissues of the crabs, thereby demonstrating trophic transfer of microplastic particles through natural predation and the ability of these particles to translocate into other tissues.

A number of studies have found instances of microplastic ingestion in other invertebrates as well, and some sampling of *in situ* organisms for microplastic ingestion has also occurred. The lugworm *Arenicola marina* has been found to ingest plastic in sediments (Thompson et al. 2004), which can impact its fitness by reducing energy reserves and cause bioaccumulation of polychlorinated biphenyls (PCBs), phenanthrene, and other pollutants (Teuten et al. 2007, Besseling et al. 2012, Browne et al. 2013). Amphipods (detritivores) and barnacles (filter feeders), despite having different feeding methods, have also been reported to ingest plastic (Thompson et al. 2004, Ugolini et al. 2013). Graham & Thompson (2009) collected four species of sea cucumber, *Thyonella gemmata, Holothuria floridana, Holothuria grisea*, and *Cucumaria frondosa*, from Florida and Maine waters to conduct microplastic feeding experiments using PVC shavings. All four species were found to preferentially ingest significantly more plastic fragments in sediments, rather than sand grains or other particles, than predicted based on the plastic to sand grain ratio. *Nephrops norvegicus*, the commercially important Norway lobster, consumes synthetic fibers in the environment that remain lodged in its stomach (Murray & Cowie 2011). Feeding experiments with *N. norvegicus* were also done using fish seeded with blue PP fibers, and 100% of lobsters had fibers in their stomachs 24 hours later. These studies

examined the potential negative effects of microplastic consumption, however, experiments performed by Kaposi et al. (2013) showed minimal effects of microplastic ingestion on the fitness of invertebrate larvae. Similarly, Goldstein and Goodwin (2013) opportunistically sampled floating debris in the Pacific Gyre and found that the rafting communities of gooseneck barnacles there frequently ingest plastic particles with no obvious ill effects.

Studies that have examined microplastic ingestion in marine vertebrates are historically relatively rare, but are now being conducted more frequently. Selective consumption of opaque white spherules occurred in 8 out of 14 fish species and a chaetognath species sampled in the Niantic Bay area by Carpenter et al. (1972). The organisms examined had ingested at least one microplastic spherule and included *Myoxocephalus aenus* (grubby), *Pseudopleuronectes americanus* (winter flounder), *Roccus americanus* (white perch), *Menidia menidia* (Atlantic silverside), *Tautogolabrus adspersus* (cunner), *Clupea harengus* (Atlantic herring), *Pollachius virens* (pollack), *Prionotus evolans* (striped searobin), and the chaetognath *Sagitta elegans*. Boerger et al. (2010) found plastic particles in the stomachs of planktivorous fish from the North Pacific Central Gyre, and Lusher et al. (2013) found plastic in the gastrointestinal tracts of five pelagic species and five demersal species of fish sampled from the English Channel. Five out of seven fish species in the North Sea, including *C. harengus*, *Merlangius merlangus* (whiting), *Gadus morhua* (cod), *Melanogrammus aeglefinus* (haddock), and *Trachurus trachurus* (horse mackerel) were found to have at least one microplastic fragment in their stomachs, more frequently in the southern half of the North Sea and English Channel than the northern half (Foekema et al. 2013). The Goiana Estuary in Brazil is contaminated with microplastic nylon fibers from fishing gear, which are sometimes consumed by two species of drum (*Stellifer brasiliensis* and *Stellifer stellifer*) in the estuary (Dantas et al. 2012). In addition to fish, seabirds

have been found with ingested plastic in their stomachs since at least the early 1960's (Rothstein 1973). Plastic is now so commonly found in seabirds that examining their gut contents has been suggested as an inexpensive way to monitor macro- and microplastic abundances in the marine environment (Ryan et al. 2009). From all of these studies examining interactions between microplastics and organisms, it can be inferred that most if not all marine organisms are at risk of the negative effects from microplastic contamination in their environment, especially given the diversity of organisms shown to be able to adsorb or ingest microplastics and the evidence of microplastic retention during trophic transfer.

Microplastic-Pollutant Interactions

Plastics can adsorb and leach out environmental pollutants while in the marine environment due to their hydrophobic structure and because of compounds added during the manufacturing process (Teuten et al. 2009). PCBs were found in a concentration of 5 ppm on the surfaces of PS pellets collected from Long Island Sound by Carpenter et al. (1972). More recently, PS pellets were found to be both sources and sinks of multiple PAH compounds in seawater (Rochman, Manzano, et al. 2013). Mato et al. (2001) found PCBs, DDE (dichlorodiphenyldichloroethylene), a breakdown product of DDT (dichlorodiphenyltrichloroethane), and plastic additive nonylphenol (NP) on PP pellets from the Japanese coast in concentrations five to six orders of magnitude higher than the surrounding seawater. On plastic pieces from the Pacific Ocean, Rios et al. (2007) detected PCBs, DDTs, and PAHs. Four years later, Hirai et al. (2011) found plastic pieces from across the Pacific containing varying concentrations of the same compounds, in addition to bisphenol A (BPA), polybrominated diphenyl ethers (PBDEs), and NP. Teuten et al. (2007) found that the PAH

phenanthrene sorbed onto microplastic particles much more readily than natural sediments and more so onto PE than PP and PVC. A summary of POP concentrations from these studies can be found in Table 1. Also, plastic pellets have been found to adsorb both major and trace metals from the marine environment, making them more bioavailable (Ashton et al. 2010). Because plastics float, they easily can disperse these pollutants to remote areas of the oceans (Barnes 2002, Zarfl & Matthies 2010).

All of these organic compounds are known POPs and endocrine-disruptors, which have the potential to negatively impact organisms that ingest plastic particles containing high concentrations of these chemicals. To examine these effects, Koelmans et al. (2013) developed a model to simulate effects of plastic-POP interactions in marine organisms, specifically *A. marina* and PCBs. The model examined both uptake and removal of POPs to body tissue through plastic ingestion; removal is a possibility if the organism's tissues already contain POPs and clean plastic is ingested, which would adsorb POPs from the tissues onto the plastic surfaces and decrease their bioaccumulation in the tissues. The results of the model indicated that the type of plastic ingested has an impact on bioaccumulation levels, as some compounds have a higher affinity for POPs than others. It also revealed that potential bioaccumulation of POPs, PCBs in this case, can vary widely with changes in equilibrium concentrations of PCBs in the sediments, pore waters, and plastic particles, as well as the concentration of particles themselves.

Organisms have been examined with respect to microplastics and POPs *in situ* as well. Fossi et al. (2012) used phthalate concentrations on microplastics and in blubber samples from Mediterranean fin whales (*Balaenoptera physalus*) to trace ingestion of plastics by baleen whales. Phthalates were similarly studied to trace possible plastic ingestion in basking sharks (*Cetorhinus maximus*), which have similar filter feeding strategies to the fin whale (Fossi et al.

2014). The muscle tissue of the basking sharks had lower phthalate concentrations than the fin whale blubber, but the compounds were still present. The combined negative effects of microplastic particles and POPs to both fitness and health have been observed in juvenile common gobies and the Japanese medaka (Oliveira et al 2013, Rochman, Hoh, et al. 2013). The results of these studies indicate that toxic chemical compounds on plastics can and do interact with organisms in the marine environment, demonstrating some of the far-reaching effects of microplastic pollution.

Both scientists and concerned citizens are putting plastics under greater scrutiny as an environmental pollutant, but the impacts of microplastics have only just begun to be investigated in earnest. The work that has been done thus far illustrates the enormity of the problem, highlighting how microplastics are known to have entered most aquatic habitats and can have physical and chemical effects on organisms from a number of trophic levels and taxa. However, there is a need for more research to create better estimates of microplastic abundance and concentration in the oceans, to further understand the impacts on marine organisms and ecology, and to come up with viable solutions to the plastics problem for the future. A useful first step toward these goals is to look for microplastics in uninvestigated locations, to quantify unknown abundance and concentrations and determine sources if possible. Once baseline knowledge of microplastics has been established, further studies can build upon it and examine more specific effects on the environment. This study was conducted to determine this baseline information about the presence of microplastics in two previously unstudied locations and used novel analysis methods to answer some preliminary questions about microplastic concentrations in minimally researched nearshore marine environments.

Chapter 2 Quantification, Description, and Comparison of Microplastics in Shinnecock and Jamaica Bays

Purpose and Study Sites

The purposes of this study are to quantify and compare the abundance and types of microplastics in two previously unstudied locations and to establish probable sources of microplastics in each bay. Investigating the prevalence of microplastics in locations as yet unexamined will add to the growing knowledge base on microplastics and help us better understand the environmental impacts of this problem. Shinnecock Bay and Jamaica Bay were chosen as the sampling locations for this study not only because they had not yet been sampled for microplastics, but also because they represent two distinctly different estuarine environments occurring on the southern shore of Long Island, both physically and in terms of human population density surrounding each bay (Figure 1).

Jamaica Bay is the western-most estuary on the south shore of Long Island (Figure 2), adjacent to New York City. It is 52.6 square kilometers and surrounded by urban development, along with three capped landfills, four wastewater treatment plants (WWTPs), and numerous combined-sewer-overflow (CSO) outputs (Benotti et al. 2007). The average water depth in Jamaica Bay is about 5 meters, but some areas are up to 20 meters deep due to dredging. Rockaway Inlet into Jamaica Bay is four times wider than the inlet into Shinnecock Bay, but Jamaica Bay has poor flushing rates and water residence times of 30-40 days due to the marsh islands that inhibit water circulation and the deep channel that has been dredged around the edge of the bay (Benotti et al. 2007). WWTPs and CSOs are the likely point source inputs for microplastics into the bay, though improperly disposed of trash from the urban environment is a

possible nonpoint source input as well. Therefore, the sites within Jamaica Bay were selected to sample as much of the bay as possible, while also targeting areas around WWTP outfalls.

Shinnecock Bay is a lagoon-type estuary on the southeastern shore of Long Island (Figure 2) enclosed by a barrier sand spit, Westhampton Dunes, with only one narrow inlet. The bay is about 33 square kilometers and has an average depth of 3 meters (Psuty & Silveira 2009). Shinnecock Inlet opens into the eastern part, while the portion of the bay on the western side of the bridge is poorly flushed. Unlike Jamaica Bay, Shinnecock Bay does not have point source wastewater inputs. The residences surrounding the bay use cesspools and septic tanks for wastewater, which slowly leaches out into the groundwater and eventually into the bay. Some possible point and nonpoint sources for microplastics into Shinnecock Bay include fishing equipment, groundwater discharge, trash improperly disposed of by recreational users, and illegal discharges from waterfront properties. The sample sites in Shinnecock Bay were chosen to obtain broad coverage of both coastal and mid-bay areas.

I hypothesized that plastics would occur in both bays but at a higher abundance in the waters of Jamaica Bay due to the larger human population and numerous point sources of wastewater. Also, I expected that the types of plastic particles found in each bay would differ based on the different environments around each bay and the point versus nonpoint sources. Types of particles that I expected included fibers in Jamaica Bay from washing machine wastewater effluent and recreational fishing, fragments in Jamaica Bay from larger pieces of debris, and fibers in Shinnecock Bay from recreational boating and fishing, which are more likely sources of fibers because of the lack of wastewater outfalls in Shinnecock Bay.

Sample Collection

Surface water samples were taken from 8 sample sites in Jamaica Bay (Figure 3) and 11 sites in Shinnecock Bay (Figure 4). The Jamaica Bay sites were chosen to target wastewater point sources of interest while also providing good coverage of the entire bay. The Shinnecock Bay sites also were chosen to provide good sample coverage of the bay, targeting both coastal and mid-bay areas. A 200 μm Sea-Gear® plankton net with a mouth 50 cm wide and equipped with a flowmeter was towed for 3 minutes at 1.5 knots at each site, sampling the top 25-50 cm of the water column. The net was positioned in the water so that the flowmeter, located in the center of the net opening, was completely submerged, but the upper edge of the net remained above the water surface. Sampling occurred from May to September 2013. Shinnecock Bay was sampled biweekly for a total of 9 dates, and Jamaica Bay was sampled once every three weeks for a total of 7 dates. Non-plastic equipment and materials were used whenever possible to avoid sample contamination. The volume-reduced water samples collected from the net were poured through a 1 mm metal sieve to remove any large algae or debris and into a glass jar. Any large particles observed were picked out of the 1 mm sieve and included in the total number of particles. Samples were stored at 8°C for up to 4 weeks until processed in the laboratory.

The glass jars were sonicated for 20 minutes at 50-60 Hz to break apart some of the biogenic material and free any plastic particles adhered to other matter. The water samples then were filtered onto a 178 μm stainless steel mesh filter 47 mm in diameter and placed in a petri dish with a tight-fitting lid. The filters were examined twice under a dissecting microscope and any plastic particles found were photographed and categorized by shape and color. Four different shapes of particles were detected: fiber, fragment, pellet, and film (Figure 5), in nine different colors: red, blue, black, white, yellow, green, orange, purple, and transparent. The

digital images were analyzed using ImageJ (Image Processing and Analysis in Java, NIH) and NISE/elements (Leica microscope software) to measure the largest dimension of each particle and separate them into size categories.

Statistical Analyses

The data were analyzed using the statistical software R (Version 3.0.2, 64-bit, The R Foundation), and the significance level for all analyses was set at $p \leq 0.05$. Only fragments and fibers were used to create size class histograms as there were limited data on pellets and films. The Kolmogorov-Smirnov (K-S) test (R code: *ks.test(data)*, $D_{n,n'} = \sup_x |F_{1,n}(x) - F_{2,n'}(x)|$ where $F_{1,n}$ and $F_{2,n'}$ are the empirical distribution functions of the two samples) was performed to compare the histogram distributions between bays.

Friedman tests were run to examine any spatial or temporal trends (R code: *friedman.test(y~A|B)*). Friedman tests are nonparametric rank order tests. In this study, Friedman tests were performed on the particle abundance data first by ranking sites within date blocks to examine potential spatial variation, then by ranking dates within site blocks to examine potential temporal variation. Friedman tests were used (e.g. instead of a two-way ANOVA (Analysis of Variance)) because the underlying particle distributions were unknown and there was no sample replication so a lack of interactions between site and date could not be assessed. Friedman tests were calculated for each bay using each of the following particle groupings: a) using all microplastic particles combined, b) using only fragments, c) using fibers only, and for each of the following blocks: d) date as the block, e) site as the block, for a total of twelve Friedman tests.

Three linear models were run comparing the distance from the sample sites in Jamaica Bay to the WWTP outfalls and to Rockaway Inlet, in order to examine possible relationships

between microplastic abundance and point sources. The first linear model compared the abundance of microplastic particles at each site to the minimum distance of the closest WWTP outfall, the second model compared abundance to the number of outfalls within 4 miles of each site, and the third model compared abundance to the distance of each site from Rockaway Inlet. CSO locations were not included as they are too numerous and are not consistent inputs.

Diversity indices from the R package *vegan* were also used. *Vegan* contains statistical indices primarily used in community ecology, so for this study each different microplastic particle color and shape combination was treated as a different "species", hereafter referred to as a "particle type", in the environment. Color was included in order to compare results with previous studies and because both shape and color can have some indication of source, such as pellets originating from industrial areas and white fibers from marina ropes. Indices used included Shannon diversity (R code: *diversity(data)*, $H = -\sum_{i=1}^{S} p_i \ln p_i$ where S is the number of particle types and p_i is the proportion of particle type i), Pielou's evenness (R code: *diversity(data)/log(specnumber(data))*, $J = \frac{H}{\log(S)}$ where H is the Shannon diversity index), and Sørensen dissimilarity index (R code: *vegdist(data, binary=TRUE)*, $\beta = \frac{a+b+c}{(2a+b+c)/2} - 1$ where a is the number of shared particle types in two sites, and b and c are the number of particle types unique to each site). Histograms of diversity and evenness were created.

Shannon diversity describes the species composition for a particular site, while Pielou's evenness indicates how evenly distributed quantities of different species are. A Shannon diversity value of 0 means that only one species is present and higher diversity values mean the species composition is more diverse. Pielou's evenness is measured from 0 to 1, and a value closer to 1 means that the quantities of different species within a sample are more evenly

distributed. Total abundance of each particle type within each sample was used to calculate both Shannon diversity and Pielou's evenness.

Sørensen dissimilarity indices have values from 0 to 1 and are used to measure beta diversity. Shannon diversity measures alpha diversity, or the diversity of species within a sample site, while beta diversity refers to the species diversity between sites or between locations. A dissimilarity index value of 0 means that all of the species between sites are shared, and a value closer to 1 means that the beta diversity is higher and the number of shared species between sites decreases. If the dissimilarity index is 1, then none of the species are shared between sites. Binary presence/absence data were used to calculate Sørensen dissimilarity indices in R to compare sites within Jamaica Bay, sites within Shinnecock Bay, and to compare Jamaica Bay as a whole and Shinnecock Bay as a whole to one another.

Quantitative and Descriptive Results

A total of 517 particles were found in the 99 samples collected from Shinnecock Bay during the five month sampling period, the majority of which were fibers (Figure 6B). The 56 samples from Jamaica Bay contained a total of 1,005 particles, which were mostly fragments (Figure 7B). Overall, 95.5% of the samples collected during this study contained at least one microplastic particle. White and blue were the most prevalent colors observed and together comprised more than 50% of the particles obtained in both bays (Figures 6A and 7A). White particles comprised 26% of the samples from Shinnecock Bay and 66% of the samples in Jamaica Bay, whereas blue particles represented 28% of the particles in Shinnecock Bay and 14% in Jamaica Bay. Red particles were more prevalent in Shinnecock Bay, making up 18% of the total as opposed to only 4% in Jamaica Bay. A few of the samples also contained

macroplastic pieces as well. The color, size, particle type, and site of all macroplastics collected are provided in Table 2.

The total concentrations of plastic particles were consistently higher at the Jamaica Bay sites than the Shinnecock Bay sites (Figure 8). In Jamaica Bay, the Jamaica Outfall site had a much higher concentration of microplastics than any of the other sites in the study, while the Canarsie Park site had a comparatively low concentration, similar to those of the Shinnecock Bay sites. Microplastic concentrations in all of the sites in Shinnecock Bay did not differ greatly from each other, implying that the particles in the surface waters there seem to be evenly distributed. Occasionally the flowmeter would not spin properly in the water, thereby resulting in inaccurate volume and particle concentration calculations. This happened to 6 samples throughout the sampling period, and these samples were excluded from the concentration calculations in Figure 8.

Digital images typical of samples from each bay show the variation in color, size, and shape of microplastic particles found during this study (Figures 9A, 9B, 10A, and 10B). Though the images demonstrate that each particle appears different and unique in each sample, there were no qualitative differences in particle types overall between bays that have not been previously discussed.

Particle size distributions in both bays were skewed to the right (Figures 11 and 12). The smallest particles observed were 200 μm, which was expected due to the sampling methodology, and the largest size observed was 5 mm. Fragment size distributions were significantly different between bays (K-S test, $d = 0.4516$, $p = 0.0036$). Jamaica Bay contained more larger-sized fragments than the Shinnecock Bay samples, accounting for the difference in means (531 μm for Shinnecock versus 615 μm for Jamaica) and medians (454μm for Shinnecock versus 498 μm for

Jamaica). Fiber size distributions were also significantly different between bays (K-S test, d = 0.3673, p = 0.0027). The Shinnecock samples had more larger-sized fibers than the Jamaica samples. The means of the two fiber histograms are very similar (1677 μm for Jamaica and 1684 μm for Shinnecock), as are the medians (1427 μm for Jamaica and 1444 μm for Shinnecock), but the general histogram shapes appear very different from each other.

There were too few pellets and films to make effective size distributions, but the particles were still photographed and measurement data were collected. The pellets from Jamaica Bay had a mean of 423 μm (N = 33), and the Shinnecock Bay pellets had a mean of 291 μm (N = 6). The films found in the Jamaica Bay samples had a mean of 2103 μm (N = 12), while the films from Shinnecock Bay had a mean of 916 μm (N = 8). On average, it appears that Shinnecock Bay samples contained larger fibers than the Jamaica Bay samples based on histogram shape, but the other three particle shapes were smaller on average than the particles in the Jamaica Bay samples.

The Shannon diversity distributions (Figure 13) are significantly different between bays (K-S test, p = 0.0154, d = 0.2608). A diversity value of 0 occurred much more frequently in the Shinnecock samples than the Jamaica samples, due to the large number of samples with either no particles or only one particle type present. Jamaica had one sample that was more diverse than any of the other samples collected (Shannon diversity index of 2.21) and contained ten different particle types. For both bays, the highest frequency of diversity values were between 1.2 and 1.5, though Jamaica Bay had equally high frequency of values between 1.5 and 1.8. This diversity value range accounts for 25% of the Shinnecock Bay samples and 23% of the Jamaica Bay samples. In general, the overall particle type diversity of any particular sample was low, usually less than five different particle types. Only one sample from the entire study had more

than ten different particle types, a sample from Jamaica Bay that contained 14, and the sample from Shinnecock Bay with the highest diversity had only seven different microplastic particle types. The sample with the highest Shannon diversity index in Jamaica Bay (2.21) had ten different particle types, not 14, but its diversity index is higher because the abundances are more evenly distributed among particle types. When grouped either by site or by date, the diversity indices did not reveal any specific site or date that had more diverse particle type composition, for either bay.

It appears that both bays had even distributions of particle types in most samples, meaning that the Pielou's evenness values were at or close to 1 (Figure 14). However, the evenness distributions are significantly different from each other (K-S test, $p = 1.36 \times 10^{-7}$, $d = 0.5007$). The few samples with a value of 0 had no microplastic particles in them, and evenness could not be calculated for the samples that contained only one particle type (included in the column "Not Calculated" in Figure 14). The high frequency of values at or close to 1 in Shinnecock Bay are due to the large number of samples containing exactly the same abundance of particles for each type in the sample. Because Shinnecock Bay had more samples that contained only a few particles, it is more likely that there would be even distribution between the few particle types present.

The beta diversity calculations in this study examined the level of dissimilarity in the compositions of microplastic particle types between sites. The Sørensen dissimilarity indices were as follows: for sites within Jamaica Bay $\beta = 0.603$, for sites within Shinnecock Bay $\beta = 0.691$, and when comparing Jamaica and Shinnecock Bays to one another $\beta = 0.170$. These results indicate that less than half of the particle types are shared between sites within each bay, but when comparing Jamaica Bay and Shinnecock Bay to one another, the microplastic beta

diversity is much lower and almost all of the particle types are shared. There were 30 different particle types found overall in Jamaica Bay, and 23 different types in Shinnecock Bay, out of the possible 36 particle types considered in this study. The samples from both bays contained high percentages of the total possible number of particle types, so inherently there would be a high degree of overlap in the particle diversities between bays.

Of the twelve Friedman tests that were run, seven had significant results. A summary of the Friedman tests conducted and their results are shown in Table 3. When examining spatial variability, Shinnecock Bay had no significant results, but the Jamaica Bay tests run using the total number of particles and using only fragments were significant ($p = 0.0033$ and 0.002, respectively). When examining temporal variability, all three tests for Shinnecock were significant (total particles: $p = 2.173 \times 10^{-5}$, only fibers: $p = 9.639 \times 10^{-5}$, only fragments: $p = 0.04$), and for Jamaica, total particles and only fragments were again significant ($p = 0.0047$ and 0.0025, respectively). The Friedman results are shown in Figures 15A, 15B, 15C, 16A, and 16B. Figures 15A, 15B, and 15C show that no particular site in Shinnecock Bay had consistently higher particle abundances than the others, hence no significant spatial differences. There does not appear to be any clear seasonal differences for Shinnecock Bay in Figures 15A, 15B, or 15C, despite having significant temporal variability according to the Friedman test. Friedman tests will return significant results if at least one pair of variables are significantly different from each other, such as August 29th and July 10th in Figure 15A, so though there may be multiple pairs of dates where the numbers of particles are different enough from each other to return a significant Friedman test result, there is no obvious seasonal pattern throughout the sampling period. In Figures 16A and 16B, the Jamaica Outfall site again appears to have much higher microplastic abundances than the other sites for almost every sampling date, similar to the concentration

results in Figure 8. This is most likely driving the significant spatial differences in Jamaica Bay's microplastic abundance. The date that seems to have generally higher abundances than the others in Figures 16A and 16B is May 29th, most likely the data responsible for the significant temporal differences indicated by the Friedman test results, however no clear seasonal pattern can be seen in the Jamaica Bay samples either.

None of the results from the linear models examining point sources scenarios in Jamaica Bay were significant. The closest to significance was the model that compared the abundance of microplastics at each site to the minimum distance to the closest WWTP outfall, with a p-value of 0.069. Though it is not definitive from the results of these linear models that WWTP outfalls are point sources of the microplastics found in the surface waters of Jamaica Bay, the results are suggestive that the abundances of microplastics in the bay can be at least partially attributed to the high volumes of wastewater that enter the bay through these WWTP outfalls.

Chapter 3 Discussion and Conclusions

Discussion

Comparison of microplastic particle characteristics between bays

While Jamaica Bay was sampled on fewer days and at a smaller number of sample sites, resulting in about half as many samples collected as for Shinnecock Bay, more microplastic particles were found in Jamaica Bay than in Shinnecock Bay in both total abundance and concentration (particles m^{-3}). White and blue were the most prevalent colors of microplastics found in this study in both bays. The abundance of white and blue particles found here is similar to the results of Gregory's (1996) and Fendall and Sewell's (2009) studies examining microplastic particles in facial cleanser products. The particles found in both studies were predominantly fragments, with some pellets, in the size range of 4.1-1240 μm and were mostly white or blue. Boerger et al.'s (2010) color characterization of microplastics ingested by fish in the North Pacific Central Gyre also found that white, clear, and blue were the colors of microplastics most frequently consumed. It could be that the white and blue particles seen, especially in Jamaica Bay, are from skincare products in household wastewater that entered the bay through the WWTPs. If facial cleansers are a large source of white microplastic particles, it could explain why Jamaica Bay, which has so much higher urban and wastewater inputs, has a larger proportion of white particles compared to Shinnecock Bay (Figures 6A and 7A). The reason for the large difference in abundance of red particles between Shinnecock and Jamaica Bays could not be determined with the sampling that was conducted.

Interestingly, the major type of microplastic particles observed differed between bays, with more fragments seen in Jamaica Bay and more fibers collected in Shinnecock Bay. It was predicted that the particle types would differ, but some of the results were unexpected, such as

the low abundance of fibers in Jamaica Bay that were predicted to be present in much higher numbers from inputs of washing machine wastewater. The large abundance of fragments in Jamaica Bay could be due to the higher input of anthropogenic trash and debris into the bay from the larger population density surrounding it, while Shinnecock Bay has fewer inputs of macroplastic debris that would be broken down into secondary microplastic fragments, but this study did not investigate this possibility in detail. Shinnecock Bay has the second largest fishing fleet in New York, located by Shinnecock Inlet, which predominantly uses trawl nets to catch fish both within the bay and in the open ocean. The greater abundance of fibers found in Shinnecock Bay could be related to the commercial fishing activity that occurs there, which is not present in Jamaica Bay. Ropes and lines on boats used to tie up to docks and used with fishing gear are most often made of synthetic material such as nylon, a preferred material because it is stronger and lasts longer than natural materials like silk or cotton. The same is true of the fishing nets themselves. Synthetic ropes and nets for commercial fishing may release microplastic fibers into the water of Shinnecock Bay and could be a major source of the fibers found there. Though the size distributions for fragments and fibers were different between the two bays, the mean lengths for each particle shape were very similar (Figures 11 and 12). In summary, we can say that microplastics from Jamaica and Shinnecock Bay were similar in terms of average size and most common colors, but differed in abundance, concentration, and dominant particle shape.

The Shannon diversities for both bays were in general very low. Though 36 different particle types were possible, most of the samples had 5 different types or less, implying that many of the particle types were very rare in the surface waters. This most likely is related to the sources of different particles. For example, if the fibers in Jamaica Bay are from washing

machine wastewater, then the diversity of colors would depend upon people's clothing color preferences, so certain colors would be much more abundant than others. Another possibility is that the colorants added to plastics may affect their buoyancy, thereby causing certain colors to sink out of the surface waters more rapidly than others. The evenness results are related to the fact that diversity was low and, for some samples, abundance was low. Some particle types were very common, and some were not seen at all, but the types that were seen frequently in these samples were mostly evenly distributed. Because of this, one specific particle type cannot be highlighted as overwhelmingly abundant.

Sørensen dissimilarity values indicated that sites within each bay have higher beta diversity and are less similar to each other than the bays as a whole are one another. Though some similarity in diversity between bays is expected, this result may seem contradictory to previously described results that illustrated the differences in particle shapes and colors between bays. The contradiction can be explained by the fact that the Sørensen index uses presence/absence data and does not take abundances into account. This means that if one particle species was found in one sample one time throughout the entire study period, it carries the same weight as a particle species that had high numbers of particles in numerous samples. Therefore, it is much easier to have more species in common when abundance is ignored, and this is why it appears as if Jamaica Bay and Shinnecock Bay have nearly identical microplastic species compositions when other results in this study have demonstrated that this is not truly the case.

The results of the Friedman tests reveal both spatial and temporal differences in the data. Runoff of microplastics into Shinnecock Bay from rainfall does not adequately explain the temporal variation in particle abundances as it had not rained on the previous day for any of the

sample dates and it was raining during sampling on only two of the dates, May 23rd and August 1st, which have lower particle abundances than some of the other dates. Rainfall could also have a dilution effect on microplastics in the surface waters instead of creating a terrestrial runoff source, especially in locations that do not have sewage or CSO inputs like Shinnecock Bay, which could explain why samples taken during or after rainfall did not have higher microplastic concentrations. This does not mean that terrestrial runoff of microplastics from rainfall is not a source of microplastics to Shinnecock Bay, but this study was unable to detect any significant effects of rainfall on microplastic particle abundance within the timeframe of the sampling period.

The lack of spatial differences in the number of particles among sites is noteworthy for Shinnecock Bay. For many other biotic and abiotic environmental factors, Shinnecock Bay has a distinct west to east gradient. Because the inlet opens into the eastern side of the bay, the western side is much less flushed, resulting in higher temperatures, lower salinities, more harmful algal blooms, higher eutrophication, lower shellfish recruitment, higher crab and predatory fish abundance, and lower pH than the eastern side (Gobler et al. 2014). Unlike any of these known west to east trends, however, the microplastic concentrations found in this study were relatively consistent throughout the surface waters and had no significant spatial variation, demonstrated by the non-significant Friedman results and the concentration results in Figure 8. This would suggest that the inlet itself is not a likely source of microplastics to the bay and that the land areas around the bay are the more likely sources, or that the input rates to Shinnecock Bay are slower than the mixing rates, making the concentrations appear the same throughout the bay. The commercial fishing fleet located near the inlet may still contribute microplastic

particles to the bay, but no distinct influence of the fleet on particle concentration was observed in the surface water samples collected during this study.

Jamaica Bay on the other hand, showed significant temporal and spatial variations in microplastic abundance for both total (all microplastic types combined) and for fragments alone (Figures 16A and 16B). In both figures, the Jamaica Outfall site had much higher abundances compared to most of the other sites. Not only is this site next to a WWTP outfall, but it is also on the opposite side of Jamaica Bay from the inlet, at a location where water would have a high residence time. The constant sewage input to this area combined with a low flushing rate may account for this large difference in microplastic abundance between this site and the others around the bay. By contrast, Rockaway is also next to a WWTP outfall, but samples from this site had much lower numbers of microplastic particles than the Jamaica Outfall site. It is much closer to the inlet though, so any particles originating from the Rockaway WWTP outfall may be more readily flushed from the bay through the inlet with the tides. These Friedman spatial test results and the linear model results suggest that the sources and variable distributions of particle abundance are more complicated and cannot be explained by WWTP outfall locations alone.

The temporal Friedman test results show differences but no clear seasonal pattern for either bay. The significant temporal variation in Jamaica Bay from the Friedman tests is most likely explained by the higher abundances of particles on May 29th than any of the other sample dates. It had rained the day before on May 28th, so terrestrial runoff and CSO inputs could be responsible for the higher abundances observed on May 29th, but there was an official CSO warning put out on July 29th because the rain on the previous day had been so heavy, yet the number of microplastics found in the samples from that date is low and very similar to the abundances from other dates, possibly due to a surface water dilution effect. This again

illustrates the complexity of microplastic sources, as well as spatial and temporal variability, and that this study cannot establish definitive point sources of microplastic particles in Jamaica Bay.

Microplastic abundance estimates vs. actual concentrations in Shinnecock and Jamaica Bays

While every effort was made to count and measure every particle in each of the samples collected, there are several factors that could have led to underestimation of microplastics in this study. There is most likely an underrepresentation of brown, green, white, and black particles, fibers especially, because these colors are more difficult to distinguish from biogenic material. Because fibers have very narrow diameters, it is possible for them to pass through the mesh filters regardless of their length, so this may be another source of underestimation of fibers. Sometimes during sampling, the plankton net caught large numbers of ctenophores and other floating debris that may have blocked the sieve and prevented some microplastic particles from going through the sieve into the sampling jars. Particle abundance could also be underestimated because some of the filters had areas with thicker layers of biogenic material that may have buried particles.

The filters were examined twice, once to count and categorize the microplastics, and again to photograph the particles for digital measurements. Not all of the particles that were observed and counted the first time could be recovered to photograph and measure, so the size histograms do not include every particle that was collected. The main reasons for this are that the fibers were highly affected by static cling, occasionally "jumping" onto the petri dish lid, and that sometimes the fragments and pellets would shift on the filter in the petri dish during transport; these factors made it difficult to relocate and recover every particle. There were also times when additional particles were found in the second examination.

Identification of the exact plastic compounds using FT-IR spectroscopy was attempted, but the particles found in this study were too small to create good quality spectra with the equipment available, so FT-IR spectral analysis was not feasible. Had identification of specific compounds been possible, that information coupled with particle shape and color could have facilitated better determination of sources. For example, if the white and blue fibers found were identified as nylon, then it is likely they are from fishing gear, as opposed to polyester or acrylic fibers that are predominantly from clothing (Browne et al. 2011). The fibers found in this study are probably polyester in Jamaica Bay and nylon in Shinnecock Bay, and the fragments in both bays are most likely PE and PP as they are currently the most common compounds used in plastic production (PlasticsEurope 2012); it therefore follows that PE and PP are also the most abundant compounds in the marine environment.

The sampling methods used in this study involved surface water tows with a plankton net and visual identification of microplastics from filtered water samples. This is a typical microplastic sampling method for collecting surface water samples. Some previously conducted studies used neuston nets, which have a rectangular opening, instead of a plankton net, and occasionally a manta net (Reisser et al. 2013) or bongo net (Doyle et al. 2011) was used. Most studies up until this point have used nets with mesh sizes 300 μm or larger, but in this study a mesh size of 200 μm was used to target smaller size classes than had previously been examined. Some plastic compounds are buoyant in water, such as polyethylene and polypropylene, and some are not, such as polystyrene. Regardless, plastics tend to accumulate biofilms and become fouled by marine organisms, changing their buoyancy and causing them to eventually sink out of the surface waters (Lobelle & Cunliffe 2011). Thompson et al. (2004) found similar plastic polymer types in water and sediment samples, implying that density is not solely responsible for

the distributions of microplastics observed. This implies that microplastics in the surface waters may represent only a small portion of the total amount of plastic particles in an ecosystem, especially in coastal and shallow eutrophic environments where plastic particles can sink to the sediments or be caught on shorelines in marshes or mangroves (Nor & Obbard 2014). Therefore, the surface water sampling methods used here may underestimate the actual concentrations of microplastics in these two bays.

Microplastic characteristics in Shinnecock and Jamaica Bays compared with other areas

The concentrations of microplastics measured in Shinnecock and Jamaica Bays ranged from 0 to 0.58 items m^{-3} and 0 to 3.93 items m^{-3}, respectively. Compared to concentrations reported in other microplastics studies conducted around the world, the concentrations of microplastics in these two bays span the low to moderate range for all locales observed (Table 4). Microplastic concentrations in Shinnecock and Jamaica Bays were quite similar to values reported for other water bodies in the U.S. Northeast region. The studies that measured microplastics in water all used surface net tows, except for Ng and Obbard (2006) who used a rotating drum sampler.

The shapes, colors, and sizes of microplastic particles found in other studies are comparable to those found here. Because microplastics are defined by size, the sizes of particles are similar in every sampling study, though some sampling and processing methods are able to collect particles an order of magnitude smaller than the plastic particles in this study. No minimum size for microplastics has been defined yet, but 1 μm particles have been found in other studies. Microplastics have been categorized by color in over 24 different studies; white is often the most common color identified (Hidalgo-Ruz et al. 2012), similar to the results of this study. The shapes of microplastics defined here (fragments, fibers, pellets, and films) were

chosen because they are common sorting categories that have been used in previous studies. Pellets and spherules have been collected in some of the earliest studies sampling microplastics (Carpenter et al. 1972, Colton et al. 1974, Austin & Stoops-Glas 1977). More recent studies have categorized microplastics into either the same shapes used here, or equivalent categories such as "granules" instead of fragments (Costa et al. 2010, Frias et al. 2010, Claessens et al. 2011, Doyle et al. 2011, Vianello et al. 2013).

The concentrations of microplastics found *in situ* versus those used in laboratory studies to examine microplastic-organism interactions tend to greatly differ. In order to have an observable effect, the concentrations in many of the laboratory studies previously summarized were at least three to four orders of magnitude higher than concentrations found in most of the water sample collection studies that have been performed, including this one (Browne et al. 2008, Graham & Thompson 2009, von Moos et al. 2012, Wegner et al. 2012, Lee et al. 2013). This may mean that microplastic concentrations in global marine environments are not high enough to have such dramatic and negative effects on marine organisms currently, but the potential still exists for marine species to be detrimentally impacted by microplastics in the future, especially as *in situ* concentrations of microplastic particles increase.

The duration of this sampling period was five months, so an examination of trends in microplastic abundance over longer time periods was outside the scope of this study, but some inferences can still be drawn about the likely trends in microplastic abundance in these two bays over time. Even a sampling period encompassing all four seasons might show greater temporal differences than what was seen in this study. Browne et al. (2011) tested the number of fibers released into washing machine water by different articles of clothing and found that polyester fleeces release 180% more fibers than polyester blankets and shirts. Because people wear more

clothes and wash more clothing during the winter, there may be more fibers from washing machine effluent ending up in marine coastal waters in the winter. With current increases in the production of plastic products and the continual breakdown of plastics already in the marine environment, it can be expected that microplastic concentrations in Shinnecock Bay and Jamaica Bay, indeed in most places globally, will only increase with time. This study therefore could be considered an initial measurement of baseline concentrations in these locations, and future studies could examine more long-term trends of microplastics around Long Island.

Conclusions

This work is a comprehensive first look at the presence of microplastics in the surface waters of two previously unstudied marine estuaries. Nothing was known about the abundance, concentration, or types of microplastic in either Jamaica Bay or Shinnecock Bay, and this is one of the few studies that examined the issue of microplastics around Long Island or in nearshore water more generally. Additionally, this study analyzed microplastic particle concentrations in a novel way by using statistics typically employed in community ecology and treating microplastic particles as different "species" present in the environment.

Both micro- and macroplastic were found in the surface waters of both study sites. There were distinct differences in the concentration, size distributions, and diversity of plastic particles between bays, with Jamaica Bay having higher microplastic abundance and concentrations than Shinnecock Bay, as predicted. In general, fibers and fragments were much more abundant than pellets and films, and white and blue were the most frequently observed colors, accounting for more than half the particles obtained in each bay. The samples from Jamaica Bay showed both temporal and spatial variations, while Shinnecock Bay had only temporal variation. The vast

majority (95.5%) of the samples collected contained at least one microplastic particle, but compared to other locations where microplastics have been sampled, concentrations in Jamaica and Shinnecock Bays are in the low to mid-range of observations. It can be assumed however, that unless improved waste management practices are put into place, microplastic concentrations will continue to increase in marine ecosystems.

Attempts to determine point sources of microplastics in Jamaica Bay were inconclusive, but the fact that different particle compositions were seen between Jamaica Bay and Shinnecock Bay would support the idea that these particles have different sources into the bays. Jamaica Bay's sources are mostly likely the point source inputs from WWTPs and CSOs, with some nonpoint inputs from improperly disposed debris, while Shinnecock Bay's sources are most likely nonpoint inputs from the surrounding land and not from the open ocean through the inlet.

Although no interactions with organisms were examined here, there is a growing amount of literature indicating that many different marine organisms can be impacted by microplastics in a variety of ways. Future microplastics work in these locations could include sampling the sediments of these two bays and examining key species *in situ* to see if they ingest or are otherwise affected by microplastics. More research also needs to be done in order to be more specific about microplastic sources to these nearshore waters. Another important aspect of future work should be further examining the possibility of trophic transfer of these microplastic particles in the environment.

Literature Cited

Aloy, A. B., Vallejo, B. M., Jr., & Juinio-Menez, M. A. (2011). Increased plastic litter cover affects the foraging activity of the sandy intertidal gastropod *Nassarius pullus*. *Mar Pollut Bull, 62*(8), 1772-1779. doi: 10.1016/j.marpolbul.2011.05.021

Andrady, A., & Neal, M. (2009). Applications and societal benefits of plastics. *Philosophical Transactions of the Royal Society B-Biological Sciences, 364*(1526), 1977-1984. doi: 10.1098/rstb.2008.0304

Andrady, A. L. (2011). Microplastics in the marine environment. *Mar Pollut Bull, 62*(8), 1596-1605. doi: 10.1016/j.marpolbul.2011.05.030

Ashton, K., Holmes, L., & Turner, A. (2010). Association of metals with plastic production pellets in the marine environment. *Mar Pollut Bull, 60*(11), 2050-2055. doi: http://dx.doi.org/10.1016/j.marpolbul.2010.07.014

Austin, H. M., & Stoops-Glas, P. M. (1977). The Distribution of Polystyrene Spheres and Nibs in Block Island Sound during 1972-1973. *Chesapeake Science, 18*(1), 89-92. doi: 10.2307/1350372

Barnes, D. K., Galgani, F., Thompson, R. C., & Barlaz, M. (2009). Accumulation and fragmentation of plastic debris in global environments. *Philos Trans R Soc Lond B Biol Sci, 364*(1526), 1985-1998. doi: 10.1098/rstb.2008.0205

Barnes, D. K. A. (2002). Biodiversity: Invasions by marine life on plastic debris. *Nature, 416*, 808+.

Benotti, M. J., Abbene, I., & Terracciano, S. A. (2007). *Nutrient Loading in Jamaica Bay, Long Island, New York: Predevelopment to 2005*. online only.

Besseling, E., Wegner, A., Foekema, E. M., van den Heuvel-Greve, M. J., & Koelmans, A. A. (2012). Effects of Microplastic on Fitness and PCB Bioaccumulation by the Lugworm Arenicola marina (L.). *Environ Sci Technol, 47*(1), 593-600. doi: 10.1021/es302763x

Betts, K. (2008). Why small plastic particles may pose a big problem in the oceans. *Environ Sci Technol*, 8995. doi: 10.1021/es802970v

Bhattacharya, P., Lin, S. J., Turner, J. P., & Ke, P. C. (2010). Physical Adsorption of Charged Plastic Nanoparticles Affects Algal Photosynthesis. *Journal of Physical Chemistry C, 114*(39), 16556-16561. doi: 10.1021/jp1054759

Boerger, C. M., Lattin, G. L., Moore, S. L., & Moore, C. J. (2010). Plastic ingestion by planktivorous fishes in the North Pacific Central Gyre. *Mar Pollut Bull, 60*(12), 2275-2278. doi: http://dx.doi.org/10.1016/j.marpolbul.2010.08.007

Browne, M. A., Crump, P., Niven, S. J., Teuten, E., Tonkin, A., Galloway, T., & Thompson, R. (2011). Accumulation of microplastic on shorelines woldwide: sources and sinks. *Environ Sci Technol, 45*(21), 9175-9179. doi: 10.1021/es201811s

Browne, M. A., Dissanayake, A., Galloway, T. S., Lowe, D. M., & Thompson, R. C. (2008). Ingested microscopic plastic translocates to the circulatory system of the mussel, Mytilus edulis (L.). *Environ Sci Technol, 42*(13), 5026-5031. doi: 10.1021/es800249a

Browne, M. A., Galloway, T. S., & Thompson, R. C. (2010). Spatial Patterns of Plastic Debris along Estuarine Shorelines. *Environ Sci Technol, 44*(9), 3404-3409. doi: 10.1021/es903784e

Browne, Mark A., Niven, Stewart J., Galloway, Tamara S., Rowland, Steve J., & Thompson, Richard C. (2013). Microplastic Moves Pollutants and Additives to Worms, Reducing

Functions Linked to Health and Biodiversity. *Current Biology, 23*(23), 2388-2392. doi: http://dx.doi.org/10.1016/j.cub.2013.10.012

Canesi, L., Ciacci, C., Fabbri, R., Marcomini, A., Pojana, G., & Gallo, G. (2012). Bivalve molluscs as a unique target group for nanoparticle toxicity. *Mar Environ Res, 76*, 16-21. doi: 10.1016/j.marenvres.2011.06.005

Carpenter, E. J., Anderson, S. J., Harvey, G. R., Miklas, H. P., & Peck, B. B. (1972). Polystyrene Spherules in Coastal Waters. *Science, 178*(4062), 749-750. doi: 10.2307/1735843

Carpenter, E. J., & Smith, K. L., Jr. (1972). Plastics on the Sargasso sea surface. *Science, 175*(4027), 1240-1241. doi: 10.1126/science.175.4027.1240

Carson, H. S. (2013). The incidence of plastic ingestion by fishes: From the prey's perspective. *Mar Pollut Bull, 74*(1), 170-174. doi: http://dx.doi.org/10.1016/j.marpolbul.2013.07.008

Carson, H. S., Colbert, S. L., Kaylor, M. J., & McDermid, K. J. (2011). Small plastic debris changes water movement and heat transfer through beach sediments. *Mar Pollut Bull, 62*(8), 1708-1713. doi: http://dx.doi.org/10.1016/j.marpolbul.2011.05.032

Choy, C. A., & Drazen, J. C. (2013). Plastic for dinner? Observations of frequent debris ingestion by pelagic predatory fishes from the central North Pacific. *MARINE ECOLOGY PROGRESS SERIES, 485*, 155-163. doi: 10.3354/meps10342

Claessens, M., De Meester, S., Van Landuyt, L., De Clerck, K., & Janssen, C. R. (2011). Occurrence and distribution of microplastics in marine sediments along the Belgian coast. *Mar Pollut Bull, 62*(10), 2199-2204. doi: 10.1016/j.marpolbul.2011.06.030

Cole, M., Lindeque, P., Fileman, E., Halsband, C., Goodhead, R., Moger, J., & Galloway, T. S. (2013). Microplastic Ingestion by Zooplankton. *Environ Sci Technol, 47*(12), 6646-6655. doi: 10.1021/es400663f

Cole, M., Lindeque, P., Halsband, C., & Galloway, T. S. (2011). Microplastics as contaminants in the marine environment: A review. *Mar Pollut Bull, 62*(12), 2588-2597. doi: http://dx.doi.org/10.1016/j.marpolbul.2011.09.025

Colton, J. B., Jr., Burns, B. R., & Knapp, F. D. (1974). Plastic particles in surface waters of the northwestern atlantic. *Science, 185*(4150), 491-497. doi: 10.1126/science.185.4150.491

Costa, M. F., Ivar do Sul, J. A., Silva-Cavalcanti, J. S., Araujo, M. C., Spengler, A., & Tourinho, P. S. (2010). On the importance of size of plastic fragments and pellets on the strandline: a snapshot of a Brazilian beach. *Environ Monit Assess, 168*(1-4), 299-304. doi: 10.1007/s10661-009-1113-4

Dantas, D. V., Barletta, M., & da Costa, M. F. (2012). The seasonal and spatial patterns of ingestion of polyfilament nylon fragments by estuarine drums (Sciaenidae). *Environmental Science and Pollution Research, 19*(2), 600-606. doi: 10.1007/s11356-011-0579-0

Davidson, T. M. (2012). Boring crustaceans damage polystyrene floats under docks polluting marine waters with microplastic. *Mar Pollut Bull, 64*(9), 1821-1828. doi: http://dx.doi.org/10.1016/j.marpolbul.2012.06.005

Derraik, J. G. B. (2002). The pollution of the marine environment by plastic debris: a review. *Mar Pollut Bull, 44*(9), 842-852. doi: 10.1016/s0025-326x(02)00220-5

Doyle, M. J., Watson, W., Bowlin, N. M., & Sheavly, S. B. (2011). Plastic particles in coastal pelagic ecosystems of the Northeast Pacific ocean. *Mar Environ Res, 71*(1), 41-52. doi: 10.1016/j.marenvres.2010.10.001

Farrell, P., & Nelson, K. (2013). Trophic level transfer of microplastic: Mytilus edulis (L.) to Carcinus maenas (L.). *Environmental Pollution, 177*(0), 1-3. doi: http://dx.doi.org/10.1016/j.envpol.2013.01.046

Fendall, L. S., & Sewell, M. A. (2009). Contributing to marine pollution by washing your face: microplastics in facial cleansers. *Mar Pollut Bull, 58*(8), 1225-1228. doi: 10.1016/j.marpolbul.2009.04.025

Foekema, E. M., De Gruijter, C., Mergia, M. T., van Franeker, J. A., Murk, A. J., & Koelmans, A. A. (2013). Plastic in North Sea Fish. *Environ Sci Technol, 47*(15), 8818-8824. doi: 10.1021/es400931b

Fossi, M. C., Coppola, D., Baini, M., Giannetti, M., Guerranti, C., Marsili, L., . . . Clò, S. (2014). Large filter feeding marine organisms as indicators of microplastic in the pelagic environment: The case studies of the Mediterranean basking shark (Cetorhinus maximus) and fin whale (Balaenoptera physalus). *Mar Environ Res*(0), in press. doi: http://dx.doi.org/10.1016/j.marenvres.2014.02.002

Fossi, M. C., Panti, C., Guerranti, C., Coppola, D., Giannetti, M., Marsili, L., & Minutoli, R. (2012). Are baleen whales exposed to the threat of microplastics? A case study of the Mediterranean fin whale (Balaenoptera physalus). *Mar Pollut Bull, 64*(11), 2374-2379. doi: http://dx.doi.org/10.1016/j.marpolbul.2012.08.013

Frias, J. P., Sobral, P., & Ferreira, A. M. (2010). Organic pollutants in microplastics from two beaches of the Portuguese coast. *Mar Pollut Bull, 60*(11), 1988-1992. doi: 10.1016/j.marpolbul.2010.07.030

Frias, J. P. G. L., Otero, V., & Sobral, P. (2014). Evidence of microplastics in samples of zooplankton from Portuguese coastal waters. *Mar Environ Res, 95*(0), 89-95. doi: http://dx.doi.org/10.1016/j.marenvres.2014.01.001

Gobler, C., Pikitch, E. K., Peterson, B., Chapman, D., Rountos, K., Santora, C., & Lowen-Klein, D. (2014). Shinnecock Bay Restoration Program Year Two Report to Doners (pp. 44). Unpublished manuscript, School of Marine and Atmospheric Sciences, Stony Brook University, Stony Brook, New York: Institute for Ocean Conservation Science.

Goldstein, M. C., & Goodwin, D. S. (2013). Gooseneck barnacles (Lepas spp.) ingest microplastic debris in the North Pacific Subtropical Gyre. *PeerJ, 1*, e184. doi: 10.7717/peerj.184

González Carman, V., Acha, E. M., Maxwell, S. M., Albareda, D., Campagna, C., & Mianzan, H. (2014). Young green turtles, Chelonia mydas, exposed to plastic in a frontal area of the SW Atlantic. *Mar Pollut Bull, 78*(1–2), 56-62. doi: http://dx.doi.org/10.1016/j.marpolbul.2013.11.012

Gouin, T., Roche, N., Lohmann, R., & Hodges, G. (2011). A Thermodynamic Approach for Assessing the Environmental Exposure of Chemicals Absorbed to Microplastic. *Environ Sci Technol*. doi: 10.1021/es1032025

Graham, E. R., & Thompson, J. T. (2009). Deposit- and suspension-feeding sea cucumbers (Echinodermata) ingest plastic fragments. *Journal of Experimental Marine Biology and Ecology, 368*(1), 22-29. doi: http://dx.doi.org/10.1016/j.jembe.2008.09.007

Gregory, M. R. (1996). Plastic 'scrubbers' in hand cleansers: A further (and minor) source for marine pollution identified. *Mar Pollut Bull, 32*(12), 867-871. doi: 10.1016/s0025-326x(96)00047-1

Habib, D., Locke, D. C., & Cannone, L. J. (1998). Synthetic fibers as indicators of municipal sewage sludge, sludge products, and sewage treatment plant effluents. *Water Air and Soil Pollution, 103*(1-4), 1-8. doi: Doi 10.1023/A:1004908110793

Hirai, H., Takada, H., Ogata, Y., Yamashita, R., Mizukawa, K., Saha, M., . . . Ward, M. W. (2011). Organic micropollutants in marine plastics debris from the open ocean and remote and urban beaches. *Mar Pollut Bull, 62*(8), 1683-1692. doi: http://dx.doi.org/10.1016/j.marpolbul.2011.06.004

Jantz, L. A., Morishige, C. L., Bruland, G. L., & Lepczyk, C. A. (2013). Ingestion of plastic marine debris by longnose lancetfish (Alepisaurus ferox) in the North Pacific Ocean. *Mar Pollut Bull, 69*(1–2), 97-104. doi: http://dx.doi.org/10.1016/j.marpolbul.2013.01.019

Kaposi, K. L., Mos, B., Kelaher, B. P., & Dworjanyn, S. A. (2013). Ingestion of Microplastic Has Limited Impact on a Marine Larva. *Environ Sci Technol, 48*(3), 1638-1645. doi: 10.1021/es404295e

Koelmans, A. A., Besseling, E., Wegner, A., & Foekema, E. M. (2013). Plastic as a Carrier of POPs to Aquatic Organisms: A Model Analysis. *Environ Sci Technol, 47*(14), 7812-7820. doi: 10.1021/es401169n

Law, K. L., Moret-Ferguson, S., Maximenko, N. A., Proskurowski, G., Peacock, E. E., Hafner, J., & Reddy, C. M. (2010). Plastic accumulation in the North Atlantic subtropical gyre. *Science, 329*(5996), 1185-1188. doi: 10.1126/science.1192321

Lee, K.-W., Shim, W. J., Kwon, O. Y., & Kang, J.-H. (2013). Size-Dependent Effects of Micro Polystyrene Particles in the Marine Copepod *Tigriopus japonicus*. *Environ Sci Technol, 47*(19), 11278-11283. doi: 10.1021/es401932b

Lobelle, D., & Cunliffe, M. (2011). Early microbial biofilm formation on marine plastic debris. *Mar Pollut Bull, 62*(1), 197-200. doi: 10.1016/j.marpolbul.2010.10.013

Lusher, A. L., McHugh, M., & Thompson, R. C. (2013). Occurrence of microplastics in the gastrointestinal tract of pelagic and demersal fish from the English Channel. *Mar Pollut Bull, 67*(1–2), 94-99. doi: http://dx.doi.org/10.1016/j.marpolbul.2012.11.028

Mato, Y., Isobe, T., Takada, H., Kanehiro, H., Ohtake, C., & Kaminuma, T. (2001). Plastic resin pellets as a transport medium for toxic chemicals in the marine environment. *Environ Sci Technol, 35*(2), 318-324. doi: Doi 10.1021/Es0010498

McDermid, K. J., & McMullen, T. L. (2004). Quantitative analysis of small-plastic debris on beaches in the Hawaiian Archipelago. *Mar Pollut Bull, 48*(7-8), 790-794. doi: 10.1016/j.marpolbul.2003.10.017

Moore, C. J. (2008). Synthetic polymers in the marine environment: A rapidly increasing, long-term threat. *Environmental Research, 108*(2), 131-139. doi: 10.1016/j.envres.2008.07.025

Moret-Ferguson, S., Law, K. L., Proskurowski, G., Murphy, E. K., Peacock, E. E., & Reddy, C. M. (2010). The size, mass, and composition of plastic debris in the western North Atlantic Ocean. *Mar Pollut Bull, 60*(10), 1873-1878. doi: 10.1016/j.marpolbul.2010.07.020

Murray, F., & Cowie, P. R. (2011). Plastic contamination in the decapod crustacean *Nephrops norvegicus* (Linnaeus, 1758). *Mar Pollut Bull, 62*(6), 1207-1217. doi: 10.1016/j.marpolbul.2011.03.032

Ng, K. L., & Obbard, J. P. (2006). Prevalence of microplastics in Singapore's coastal marine environment. *Mar Pollut Bull, 52*(7), 761-767. doi: 10.1016/j.marpolbul.2005.11.017

O'Brine, T., & Thompson, R. C. (2010). Degradation of plastic carrier bags in the marine environment. *Mar Pollut Bull, 60*(12), 2279-2283. doi: http://dx.doi.org/10.1016/j.marpolbul.2010.08.005

Oliveira, M., Ribeiro, A., & Guilhermino, L. (2012). Effects of exposure to microplastics and PAHs on microalgae *Rhodomonas baltica* and *Tetraselmis chuii*. *Comparative Biochemistry and Physiology a-Molecular & Integrative Physiology, 163*(1), S19-S20. doi: 10.1016/j.cbpa.2012.05.062

Oliveira, M., Ribeiro, A., Hylland, K., & Guilhermino, L. (2013). Single and combined effects of microplastics and pyrene on juveniles (0+group) of the common goby *Pomatoschistus microps* (Teleostei, Gobiidae). *Ecological Indicators, 34*, 641-647. doi: 10.1016/j.ecolind.2013.06.019

PlasticsEurope. (2012). Plastics - the Facts 2012 (pp. 40).

Psuty, N. P., & Silveira, T. M. (2009). Geomorphological Evolution of Estuaries: The Dynamic Basis for Morpho-Sedimentary Units in Selected Estuaries in the Northeastern United States. *Marine Fisheries Review, 71*, 34-45.

Reisser, J., Shaw, J., Wilcox, C., Hardesty, B. D., Proietti, M., Thums, M., & Pattiaratchi, C. (2013). Marine Plastic Pollution in Waters around Australia: Characteristics, Concentrations, and Pathways. *PLoS ONE, 8*(11), e80466. doi: 10.1371/journal.pone.0080466

Rios, L. M., Moore, C., & Jones, P. R. (2007). Persistent organic pollutants carried by synthetic polymers in the ocean environment. *Mar Pollut Bull, 54*(8), 1230-1237. doi: http://dx.doi.org/10.1016/j.marpolbul.2007.03.022

Rochman, C. M., Hoh, E., Kurobe, T., & Teh, S. J. (2013). Ingested plastic transfers hazardous chemicals to fish and induces hepatic stress. *Scientific Reports, 3*. doi: 10.1038/srep03263

Rochman, C. M., Manzano, C., Hentschel, B. T., Simonich, S. L. M., & Hoh, E. (2013). Polystyrene Plastic: A Source and Sink for Polycyclic Aromatic Hydrocarbons in the Marine Environment. *Environ Sci Technol, 47*(24), 13976-13984. doi: 10.1021/es403605f

Romer, J. R. (2008). The evolution of San Francisco's plastic bag ban. *Golden Gate University Environmental Law Journal, 1*, 439-465.

Rothstein, S. I. (1973). Plastic Particle Pollution of the Surface of the Atlantic Ocean: Evidence from a Seabird. *The Condor, 75*(3), 344-345. doi: 10.2307/1366176

Ryan, P. G., Moore, C. J., van Franeker, J. A., & Moloney, C. L. (2009). Monitoring the abundance of plastic debris in the marine environment. *Philosophical Transactions of the Royal Society B: Biological Sciences, 364*(1526), 1999-2012. doi: 10.1098/rstb.2008.0207

Setälä, O., Fleming-Lehtinen, V., & Lehtiniemi, M. (2014). Ingestion and transfer of microplastics in the planktonic food web. *Environmental Pollution, 185*(0), 77-83. doi: http://dx.doi.org/10.1016/j.envpol.2013.10.013

Teuten, E. L., Rowland, S. J., Galloway, T. S., & Thompson, R. C. (2007). Potential for plastics to transport hydrophobic contaminants. *Environ Sci Technol, 41*(22), 7759-7764.

Teuten, E. L., Saquing, J. M., Knappe, D. R. U., Barlaz, M. A., Jonsson, S., Björn, A., . . . Takada, H. (2009). Transport and release of chemicals from plastics to the environment and to wildlife. *Philosophical Transactions of the Royal Society B: Biological Sciences, 364*(1526), 2027-2045. doi: 10.1098/rstb.2008.0284

Thompson, R. C., Olsen, Y., Mitchell, R. P., Davis, A., Rowland, S. J., John, A. W. G., . . . Russell, A. E. (2004). Lost at sea: Where is all the plastic? *Science, 304*(5672), 838-838. doi: 10.1126/science.1094559

Ugolini, A., Ungherese, G., Ciofini, M., Lapucci, A., & Camaiti, M. (2013). Microplastic debris in sandhoppers. *Estuarine Coastal and Shelf Science, 129*, 19-22. doi: 10.1016/j.ecss.2013.05.026

Van Cauwenberghe, L., Claessens, M., Vandegehuchte, M. B., Mees, J., & Janssen, C. R. (2013). Assessment of marine debris on the Belgian Continental Shelf. *Mar Pollut Bull, 73*(1), 161-169. doi: http://dx.doi.org/10.1016/j.marpolbul.2013.05.026

Van Cauwenberghe, L., Vanreusel, A., Mees, J., & Janssen, C. R. (2013). Microplastic pollution in deep-sea sediments. *Environmental Pollution, 182*, 495-499. doi: 10.1016/j.envpol.2013.08.013

Vianello, A., Boldrin, A., Guerriero, P., Moschino, V., Rella, R., Sturaro, A., & Da Ros, L. (2013). Microplastic particles in sediments of Lagoon of Venice, Italy: First observations on occurrence, spatial patterns and identification. *Estuarine, Coastal and Shelf Science, 130*(0), 54-61. doi: http://dx.doi.org/10.1016/j.ecss.2013.03.022

von Moos, N., Burkhardt-Holm, P., & Köhler, A. (2012). Uptake and Effects of Microplastics on Cells and Tissue of the Blue Mussel Mytilus edulis L. after an Experimental Exposure. *Environ Sci Technol, 46*(20), 11327-11335. doi: 10.1021/es302332w

Wegner, A., Besseling, E., Foekema, E. M., Kamermans, P., & Koelmans, A. A. (2012). Effects of nanopolystyrene on the feeding behavior of the blue mussel (*Mytilus edulis* L.). *Environ Toxicol Chem, 31*(11), 2490-2497. doi: 10.1002/etc.1984

Wilber, R. J. (1987). Plastic in the North-Atlantic. *Oceanus, 30*(3), 61-&.

Wright, S. L., Rowe, D., Thompson, R. C., & Galloway, T. S. (2013). Microplastic ingestion decreases energy reserves in marine worms. *Current Biology, 23*(23), R1031-R1033. doi: http://dx.doi.org/10.1016/j.cub.2013.10.068

Zarfl, C., & Matthies, M. (2010). Are marine plastic particles transport vectors for organic pollutants to the Arctic? *Mar Pollut Bull, 60*(10), 1810-1814. doi: 10.1016/j.marpolbul.2010.05.026

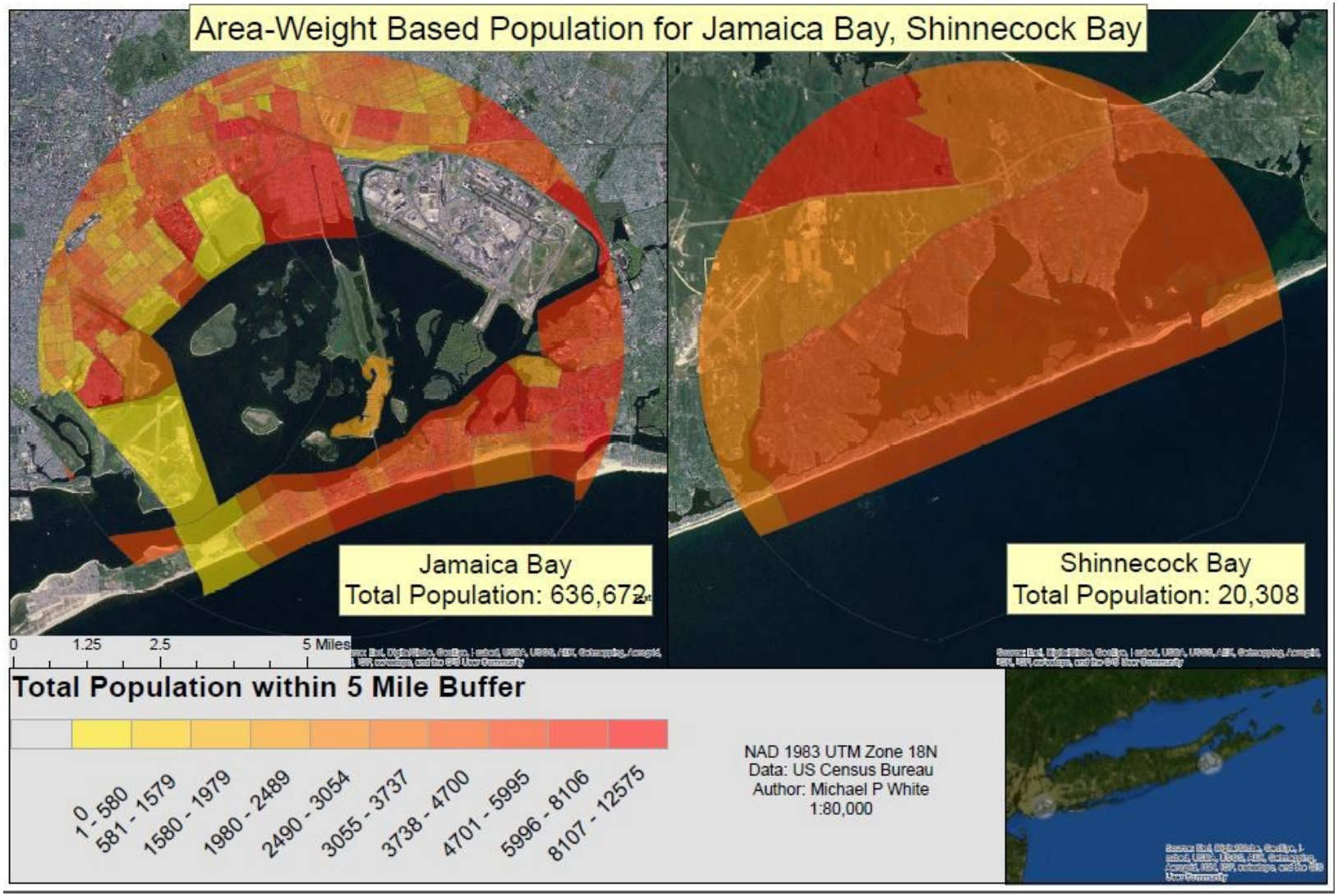


Figure 1 Population densities surrounding Jamaica Bay and Shinnecock Bay based on data from the 2010 US Census Bureau.

Figure 2 The locations of the two study sites in relation to each other and the surrounding area. Image Source: Google Earth.

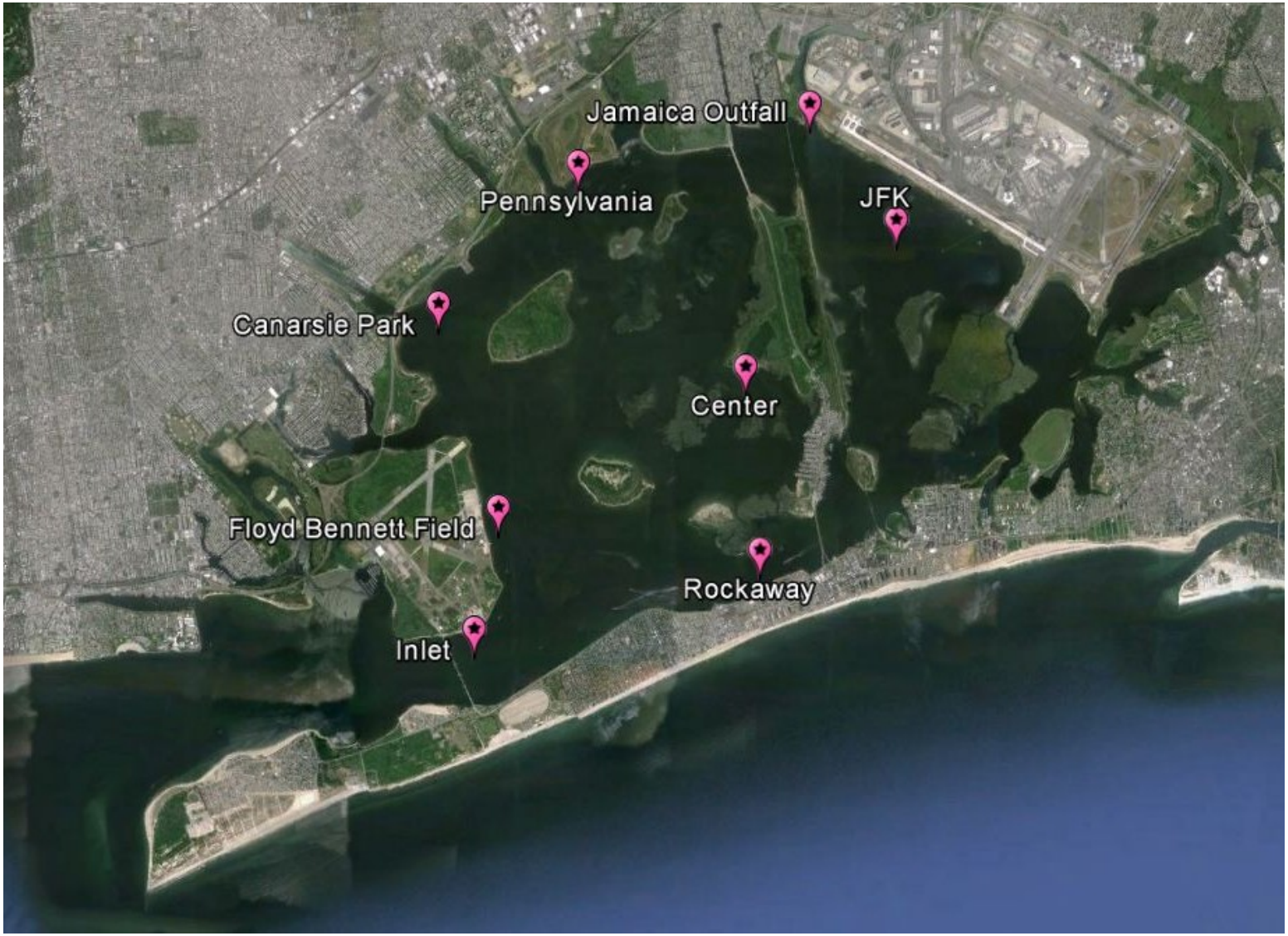


Figure 3 Map of the 8 sample sites in Jamaica Bay. Image Source: Google Earth, labels added by author.

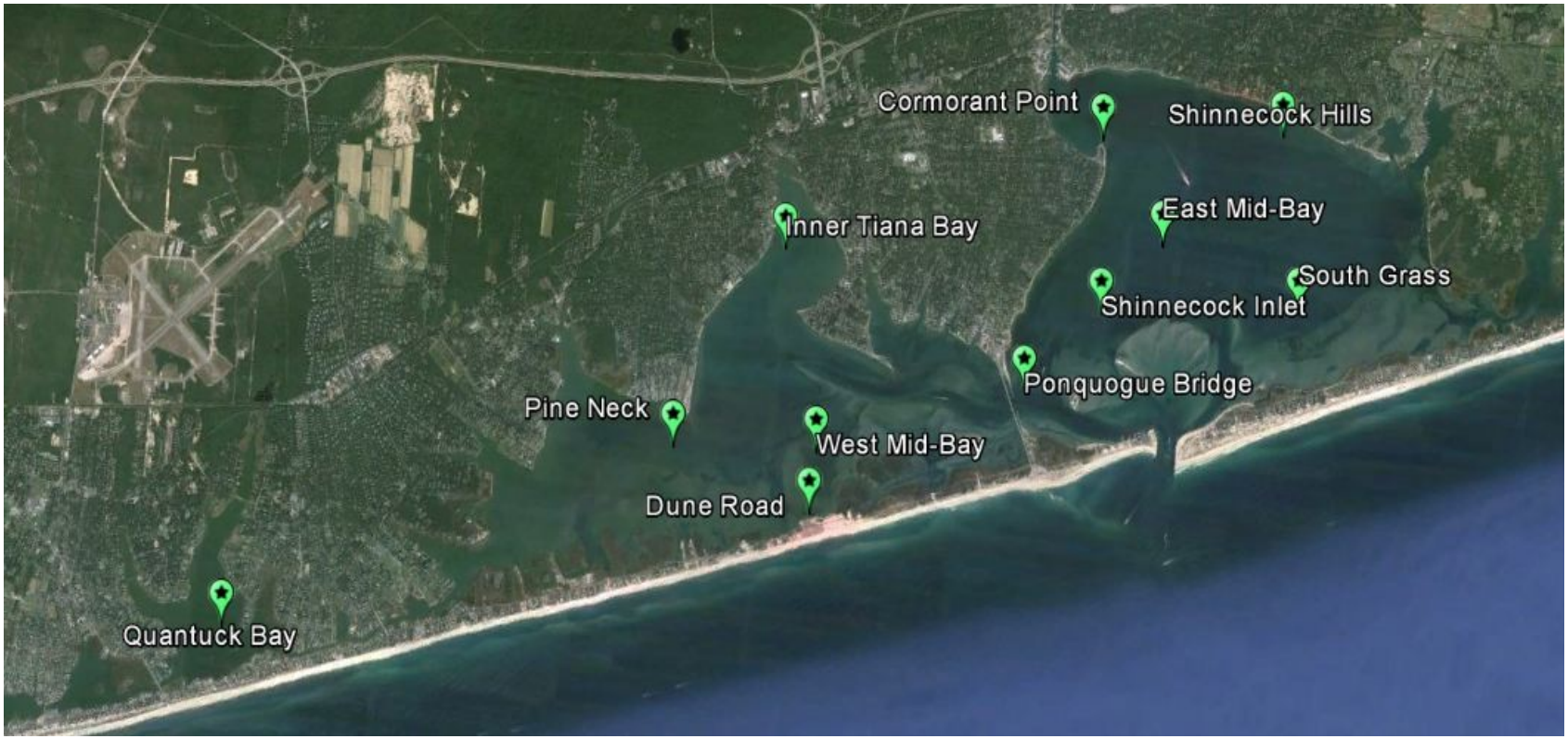


Figure 4 Map of the 11 sample sites in Shinnecock Bay. Image Source: Google Earth, labels added by author.

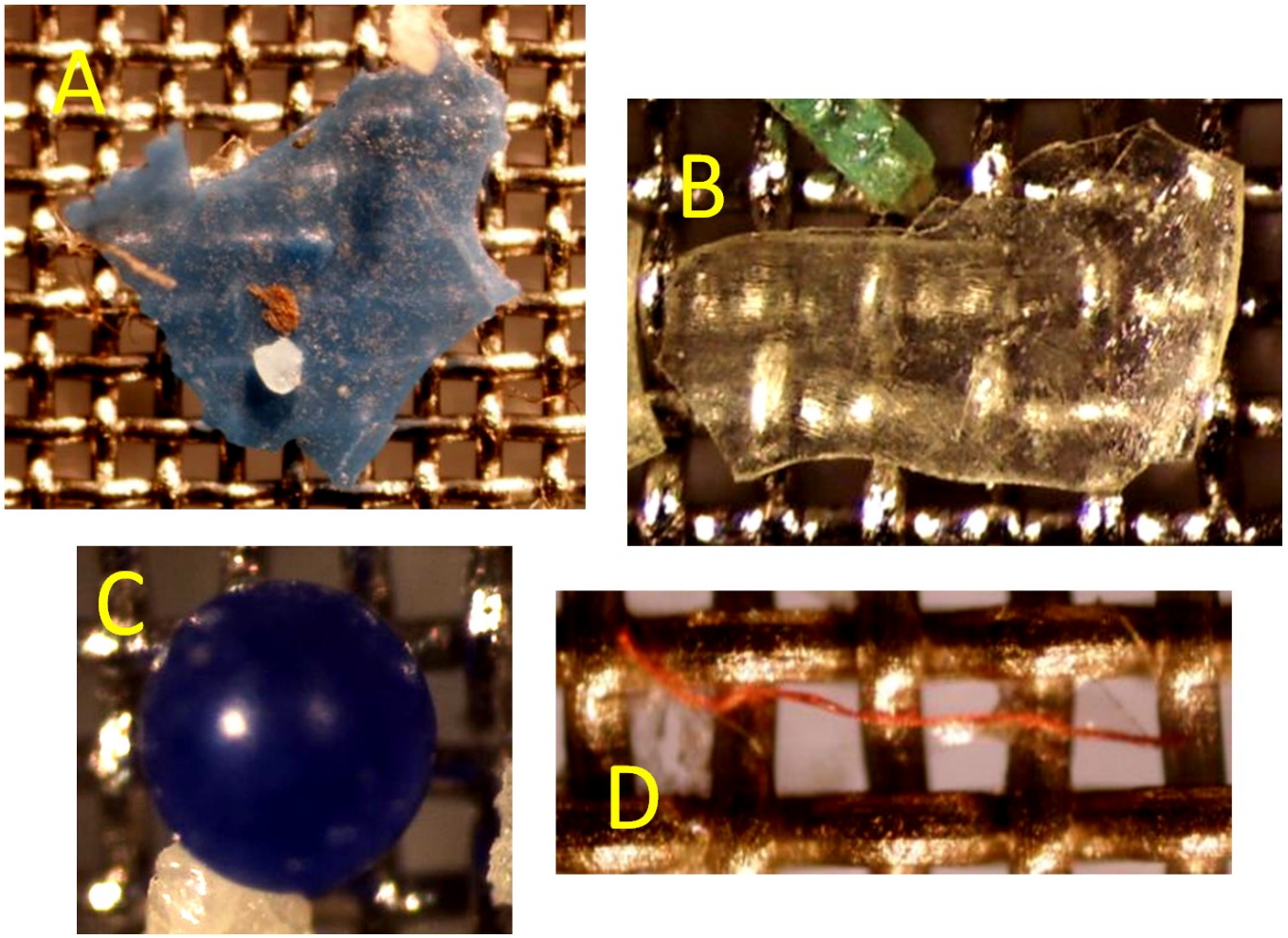


Figure 5 Example images of each particle shape. A) fragment, B) film, C) pellet, D) fiber.

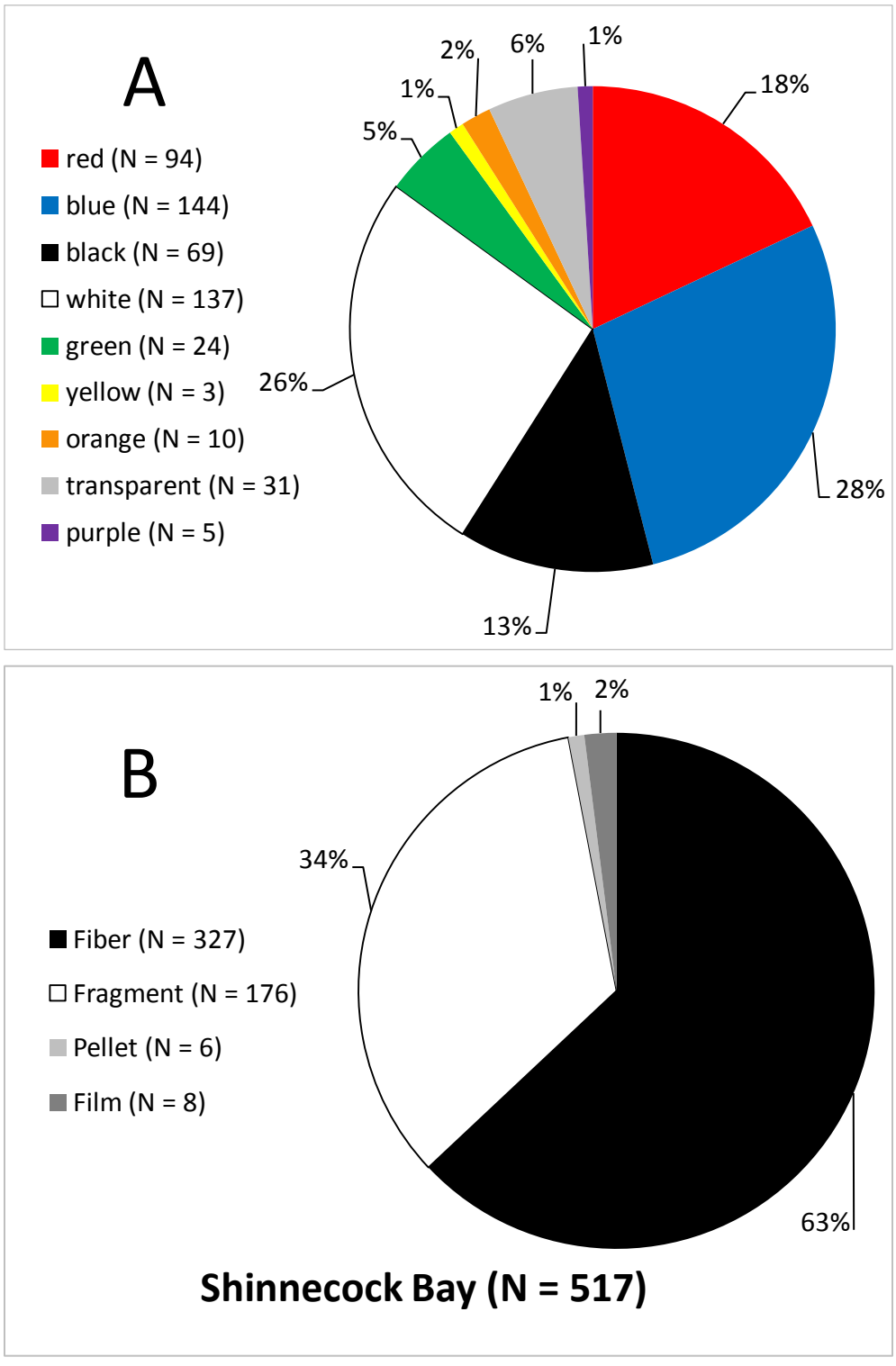


Figure 6 The percentage breakdown of microplastic particles from Shinnecock Bay. A) Particles sorted by color, B) Particles sorted by shape.

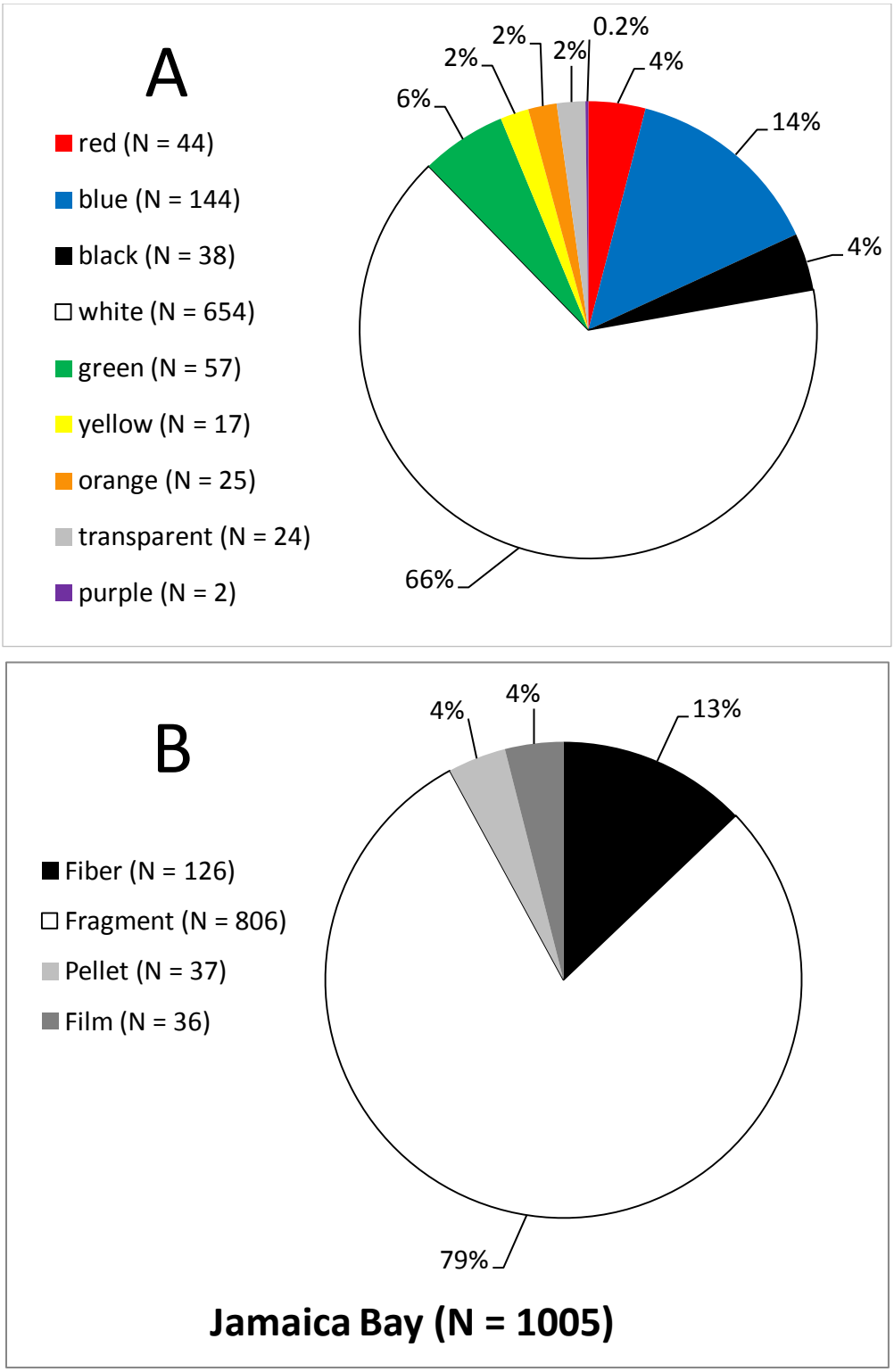


Figure 7 The percentage breakdown of microplastic particles in Jamaica Bay. A) Particles sorted by color, B) Particles sorted by shape.

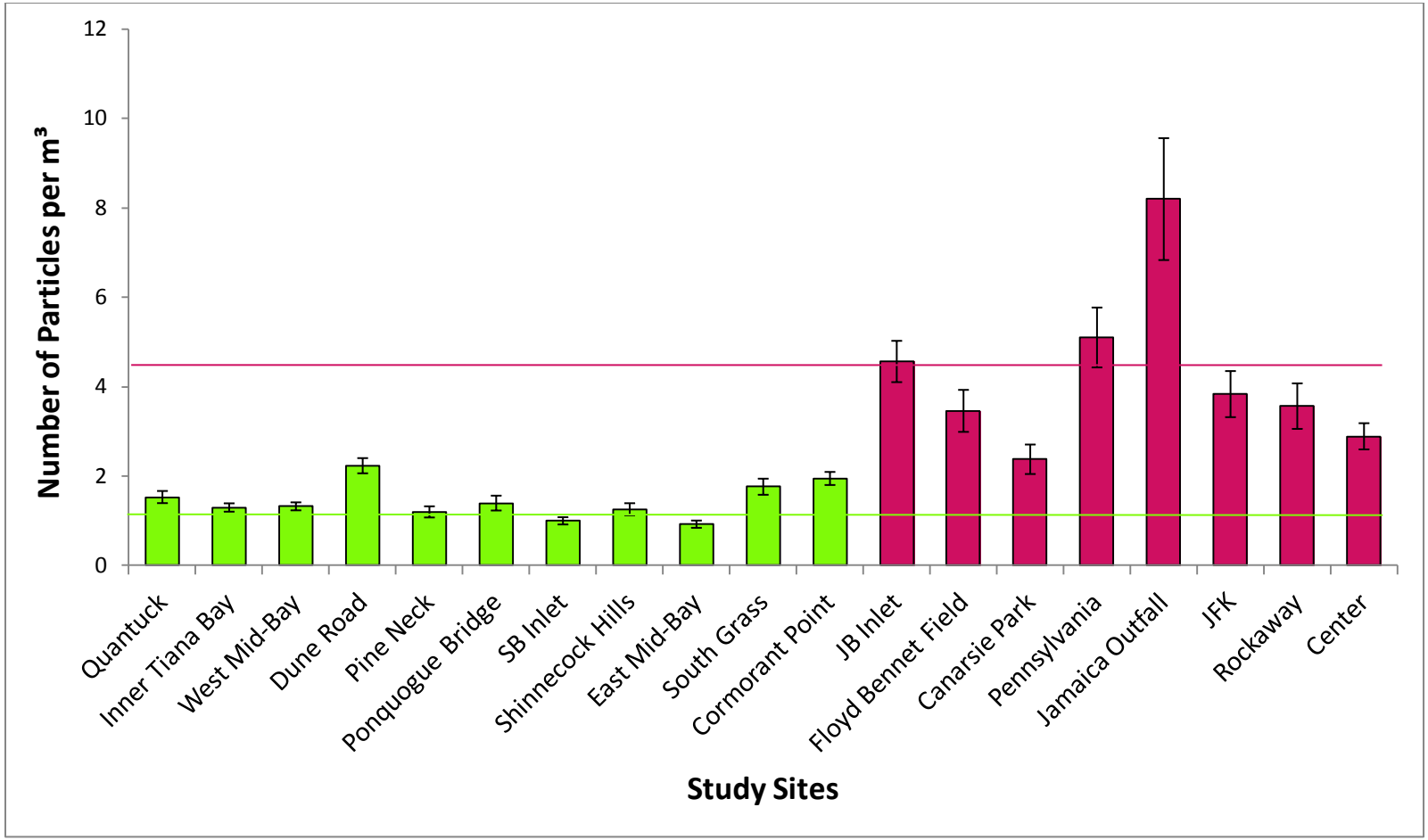


Figure 8 The total concentration of plastic particles found at each site with SD error bars, horizontal lines indicate means (1.44 items m^{-3} and 4.25 items m^{-3}). Green bars are Shinnecock sites and magenta bars are Jamaica sites.

Figure 9A Typical microplastic fragments and pellets from Jamaica Bay. Scale bar is 500 microns.

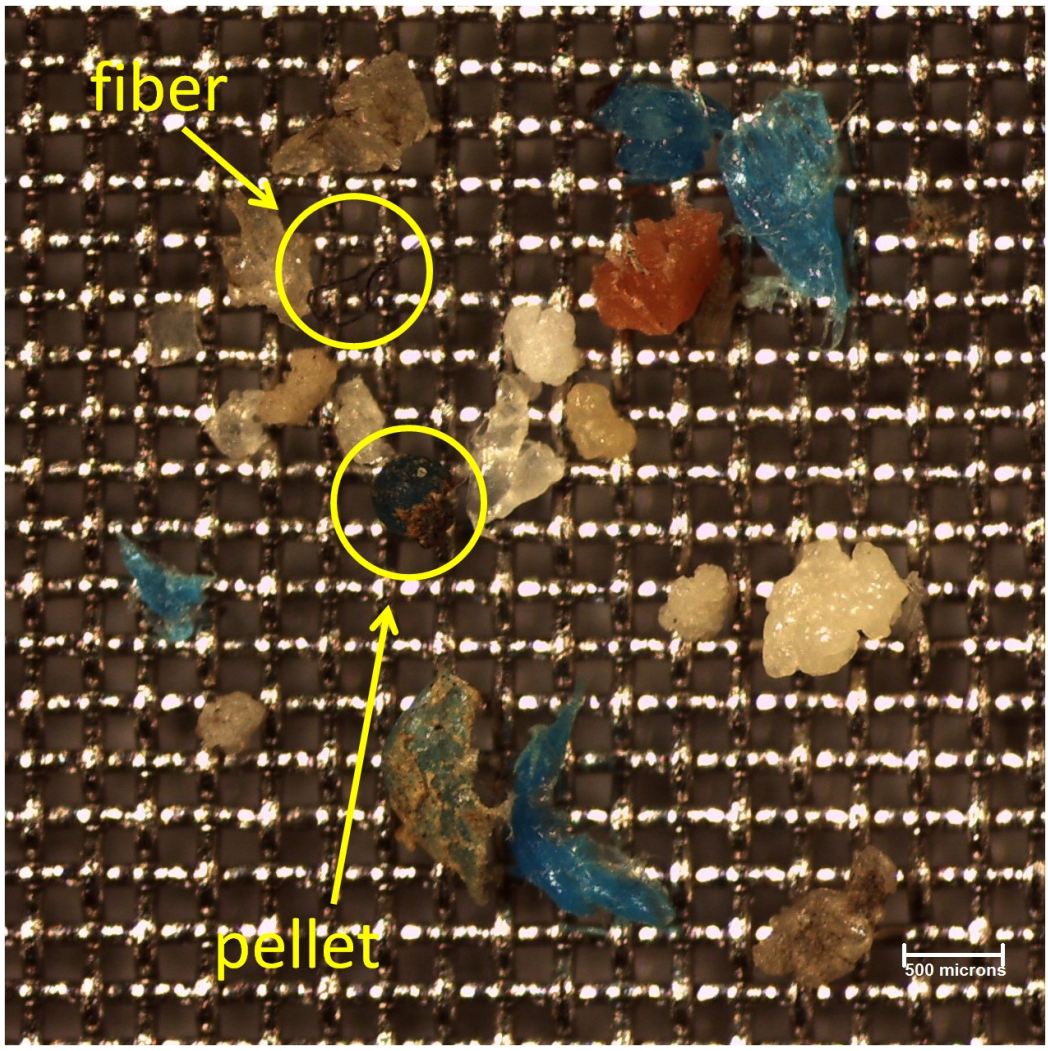


Figure 9B Typical microplastic fragments, a pellet, and a fiber from Jamaica Bay. Scale bar is 500 microns.

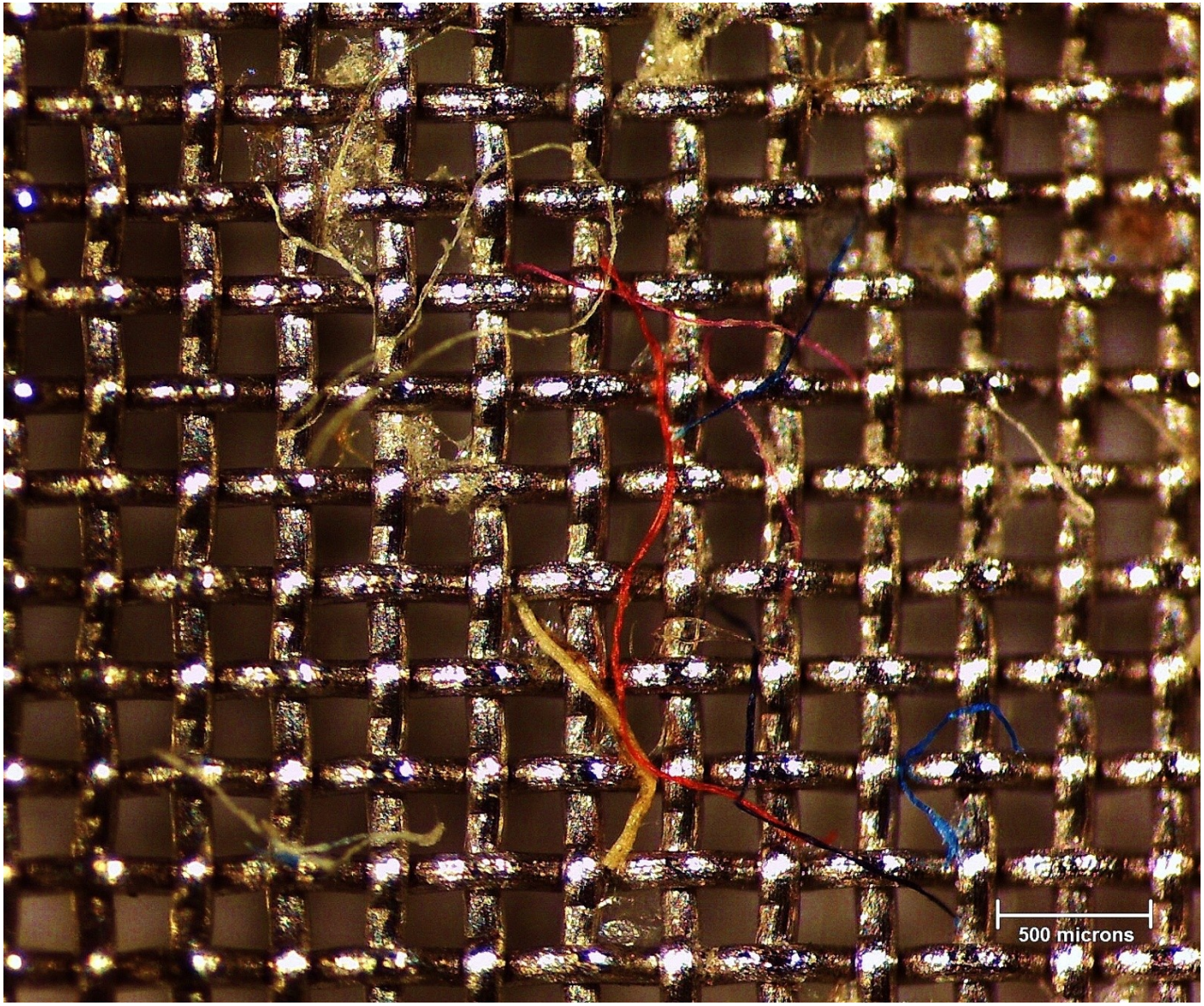


Figure 10A Typical microplastic fibers from Shinnecock Bay. Scale bar is 500 microns.

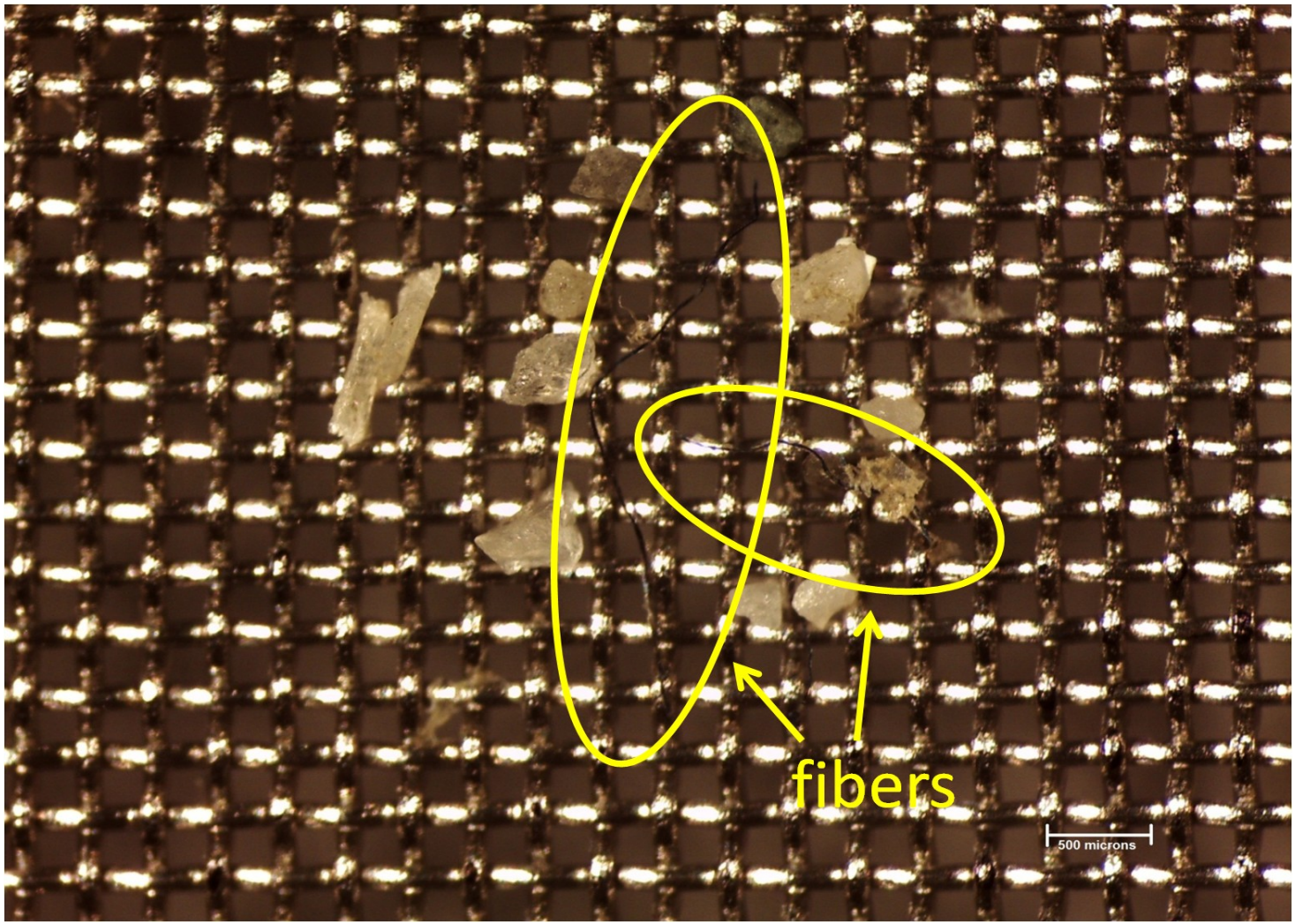


Figure 10B Typical microplastic fragments and fibers from Shinnecock Bay. Scale bar is 500 microns.

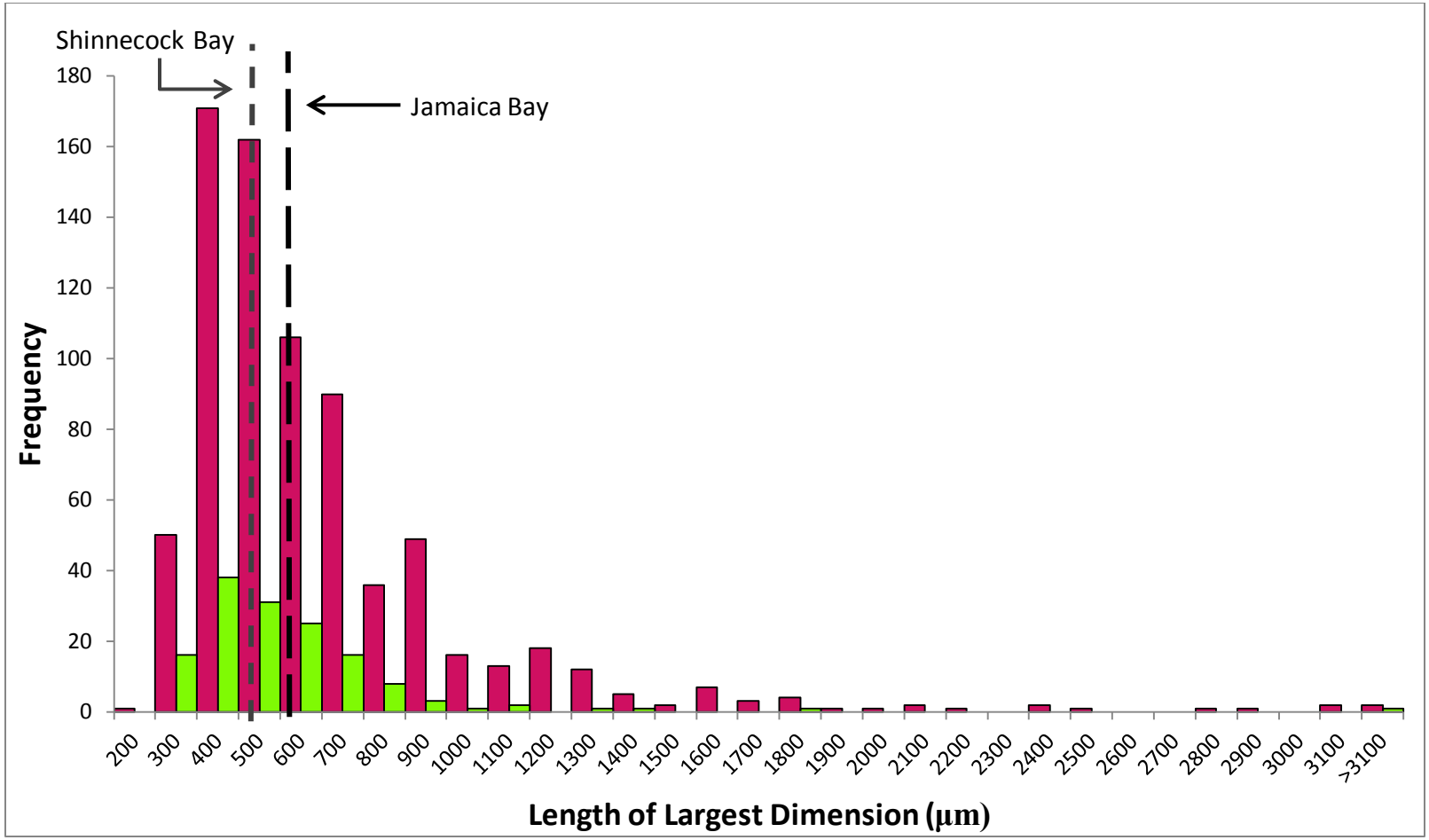


Figure 11 Size distribution of fragments in samples from Jamaica Bay (N=759) in magenta and from Shinnecock Bay (N = 144) in green. Column >3100 for Jamaica contains two particles, one 4365.59 µm and the second 4806.06 µm, and for Shinnecock contains one particle 4993.81µm. Dashed lines represent the means (531 µm and 615 µm), and the medians are 454µm for Shinnecock and 498 µm for Jamaica.

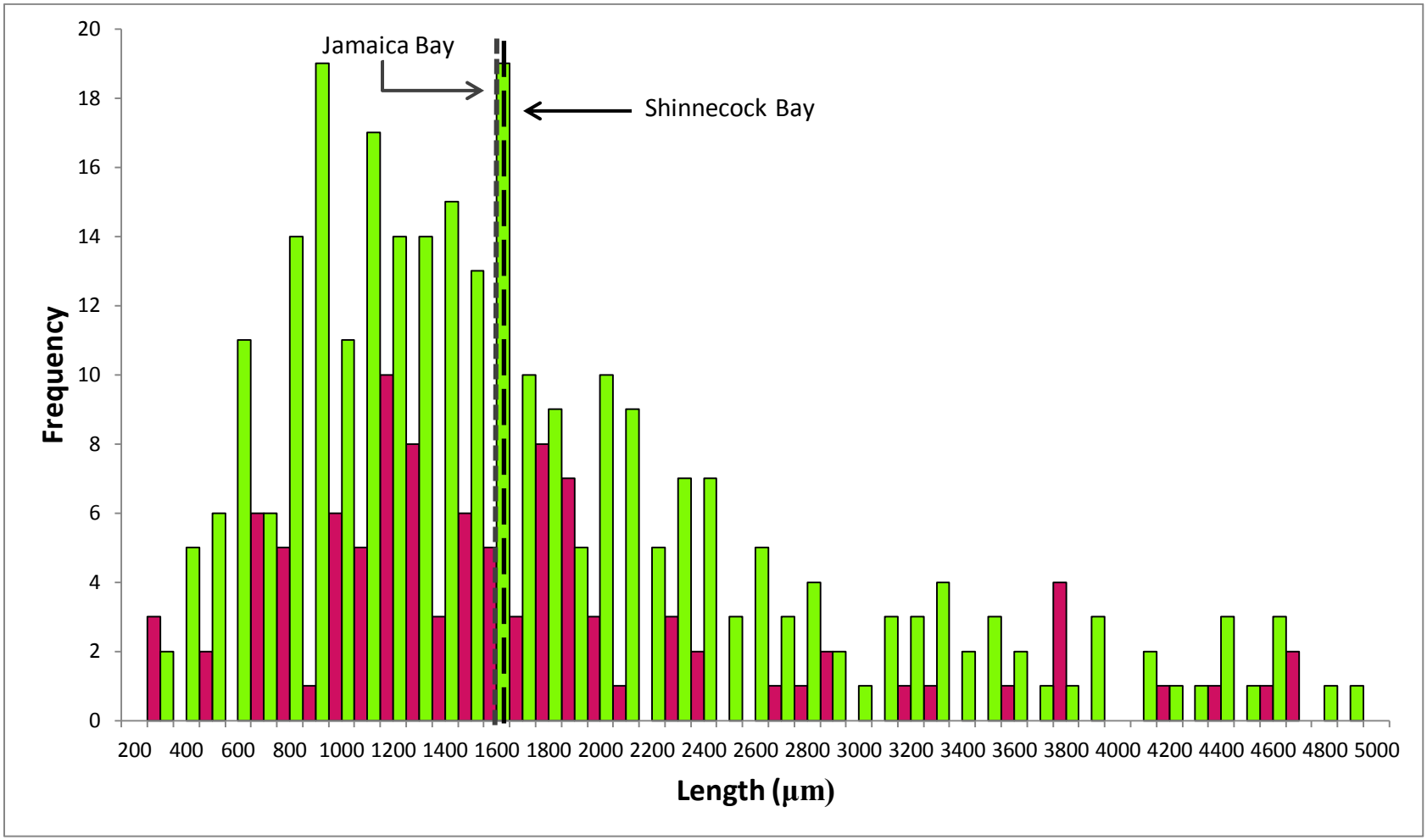
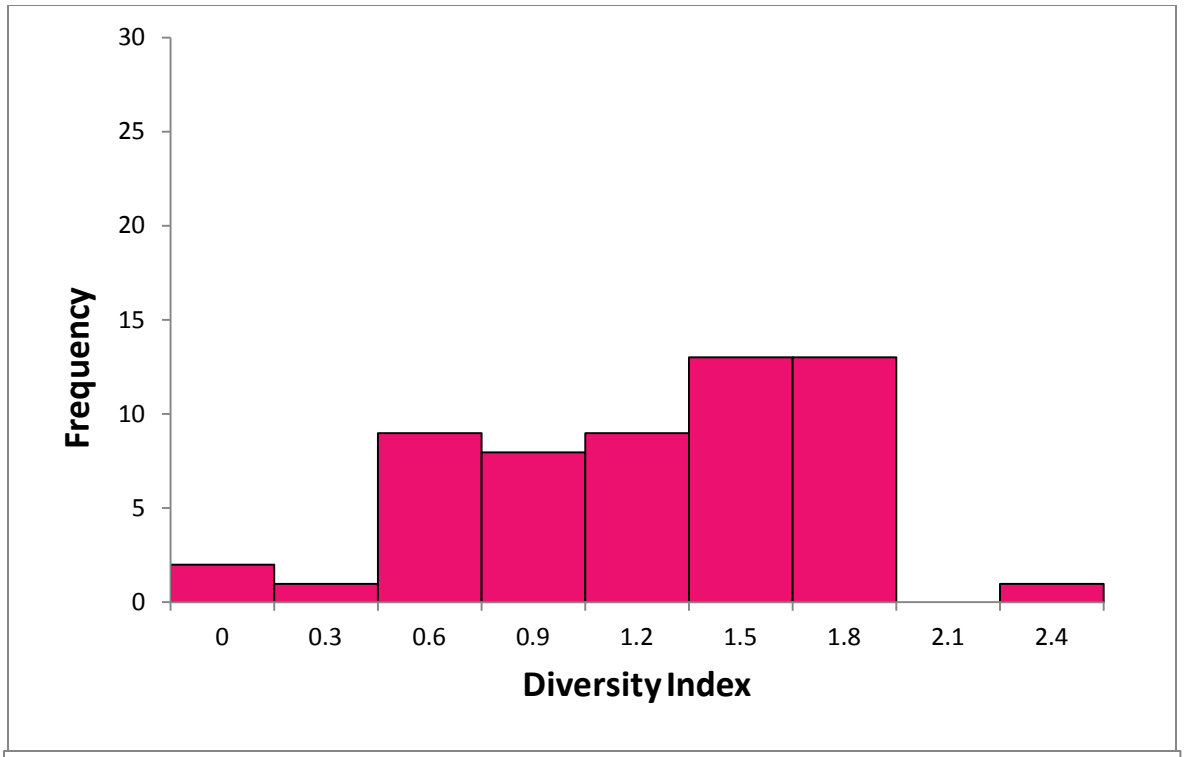


Figure 12 Size distribution of fibers in samples from Jamaica Bay (N = 103) in magenta and Shinnecock Bay (N = 281) in green. Dashed lines represent the means (1677 µm and 1684 µm), and the medians are 1427 µm for Jamaica and 1444 µm for Shinnecock.



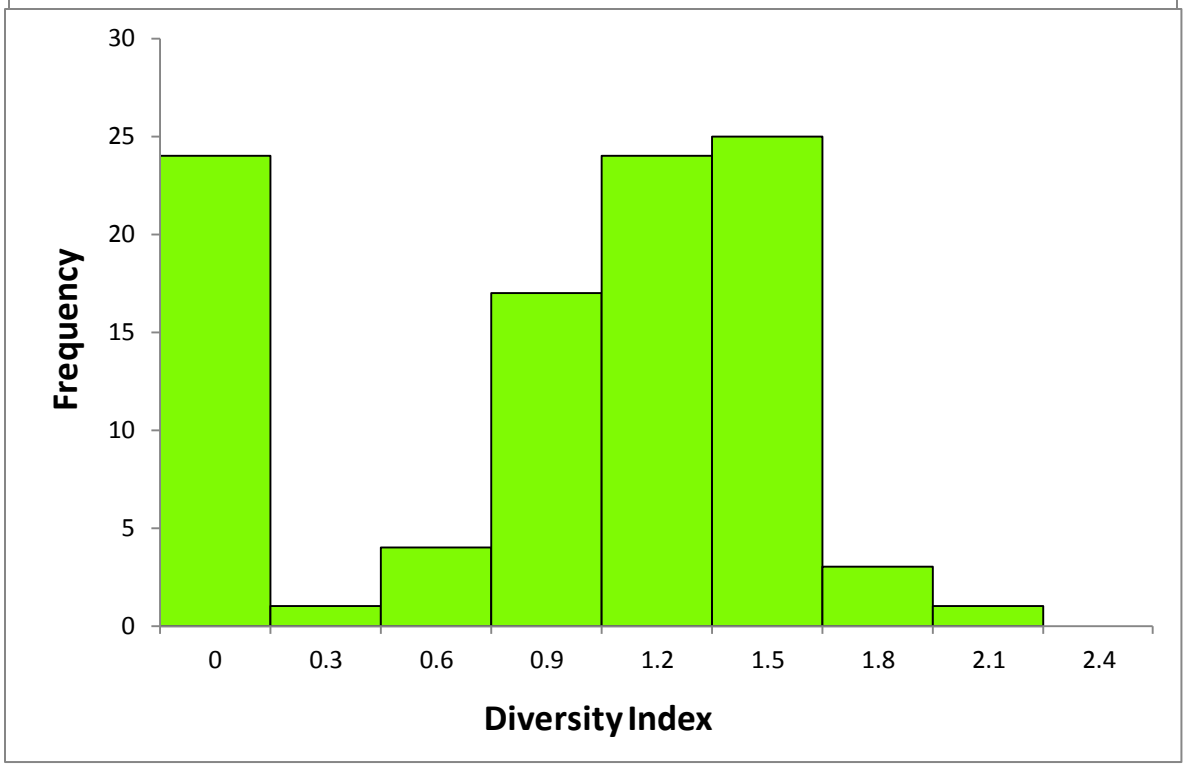


Figure 13 Shannon diversity indices as histograms, Jamaica Bay on the top and Shinnecock Bay on the bottom.

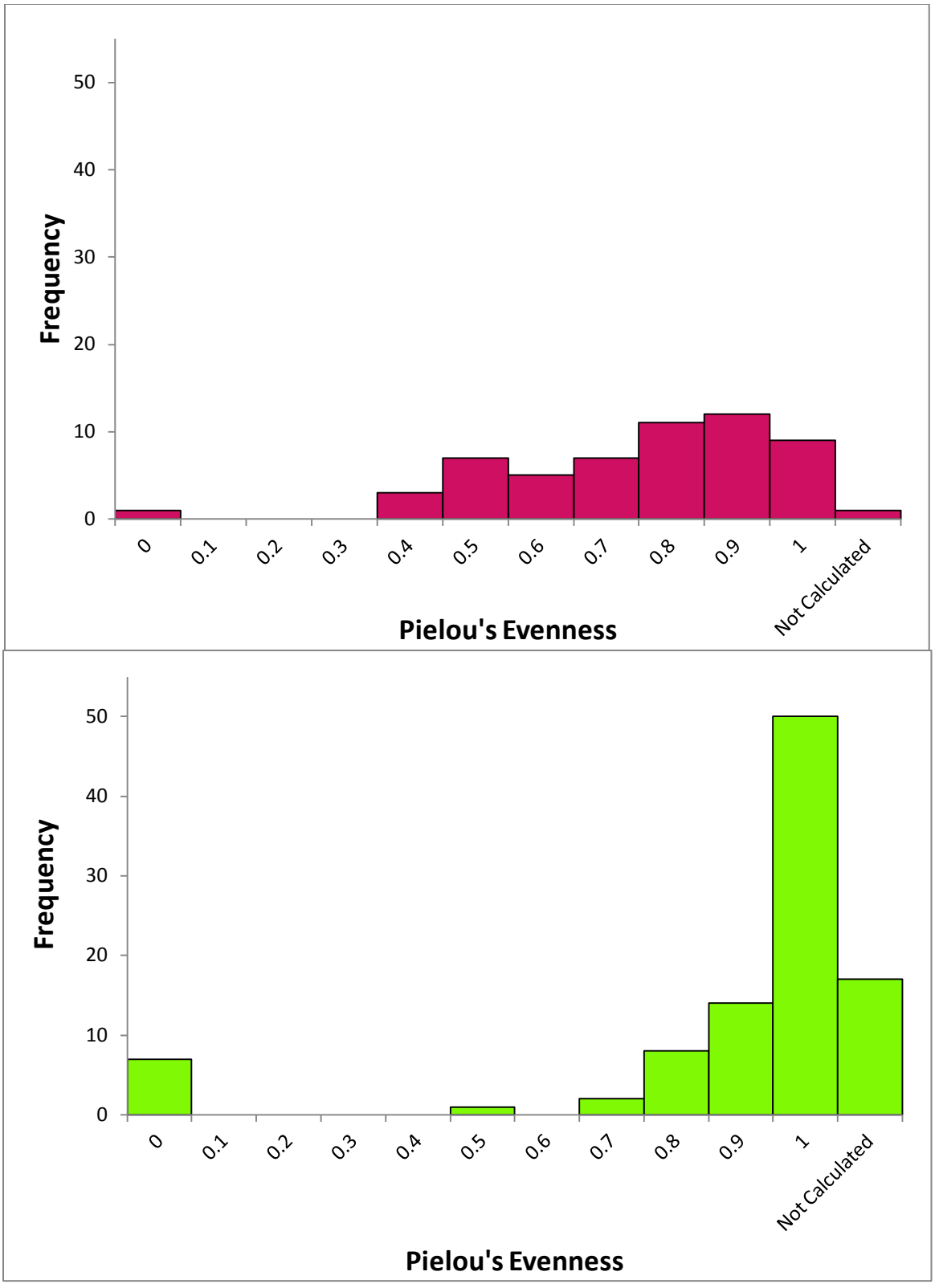


Figure 14 Histograms of Pielou's evenness, Jamaica Bay on top and Shinnecock Bay on the bottom.

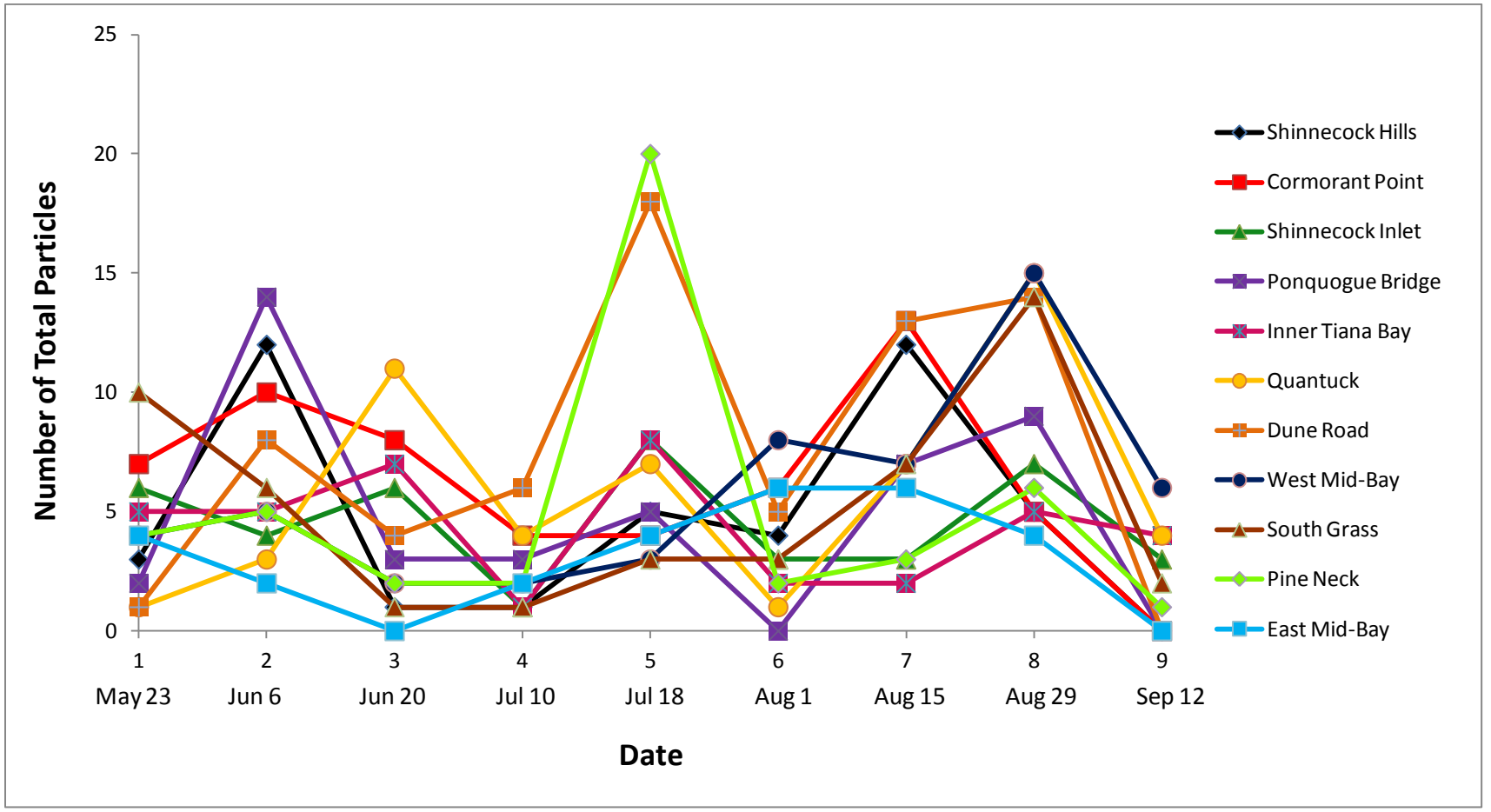


Figure 15A Friedman test results comparing the number of total particles to sites and dates sampled for Shinnecock Bay.

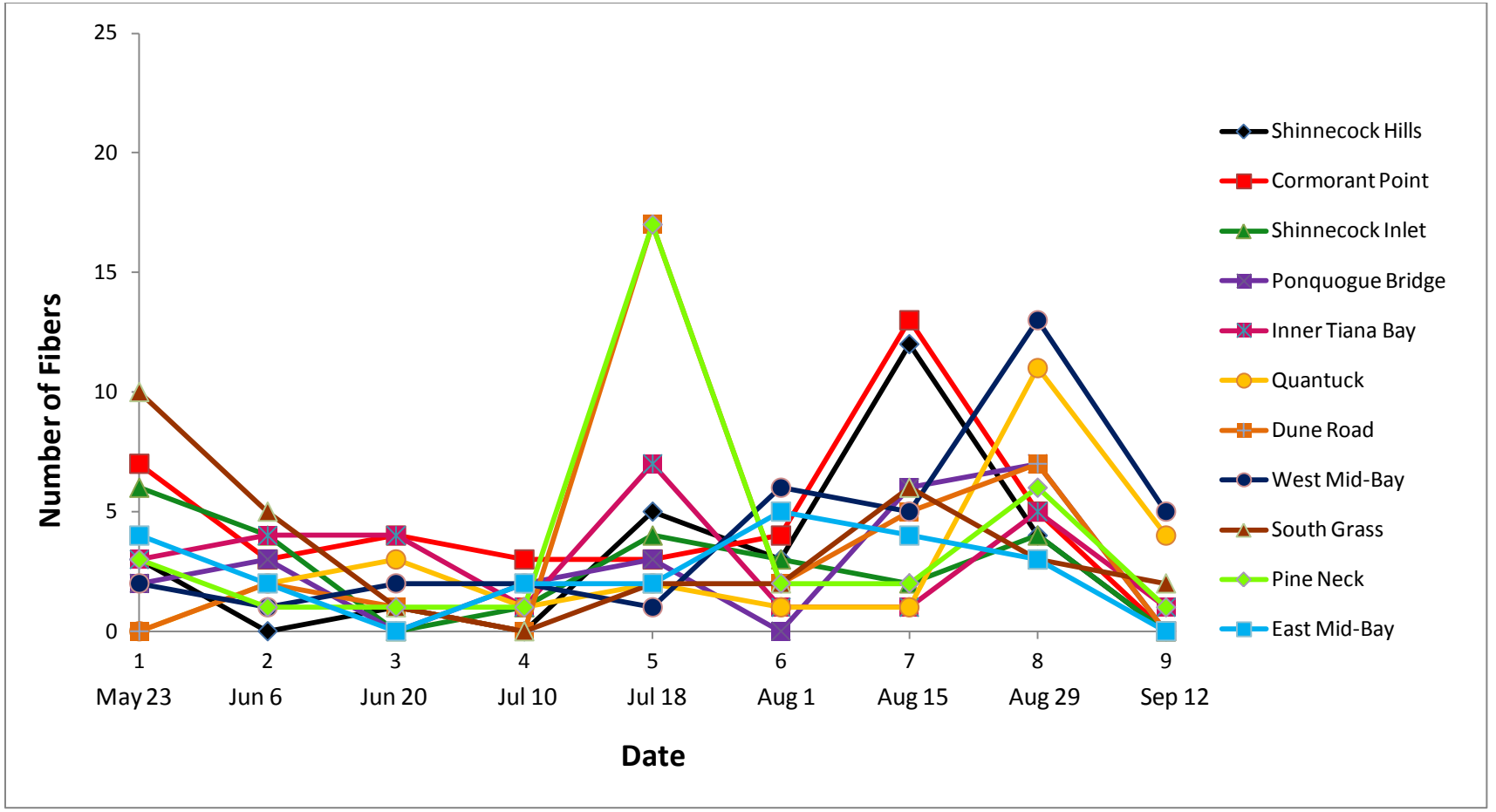


Figure 15B Friedman test results comparing fibers to sites and dates sampled for Shinnecock Bay.

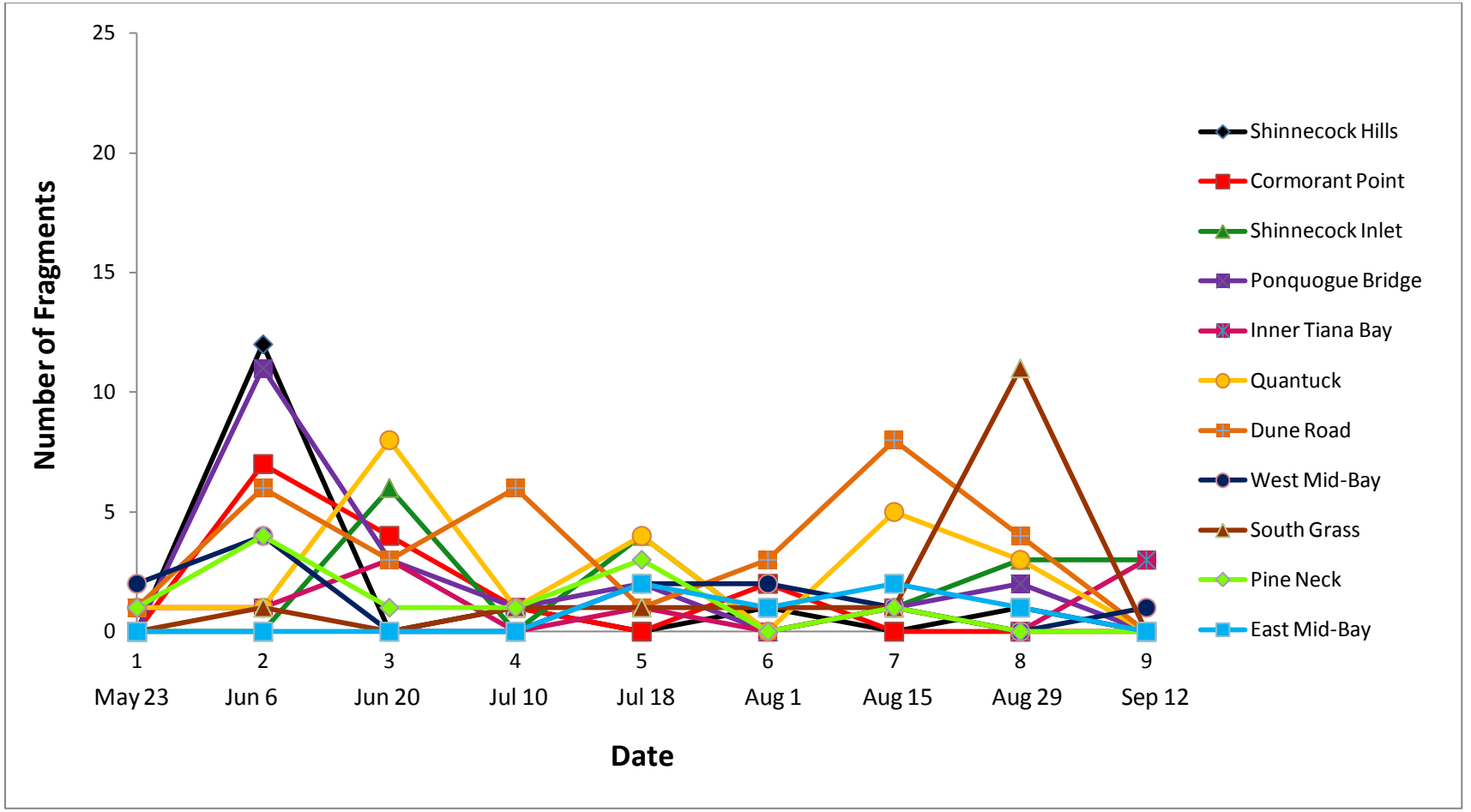


Figure 15C Friedman test results comparing fragments to sites and dates sampled for Shinnecock Bay.

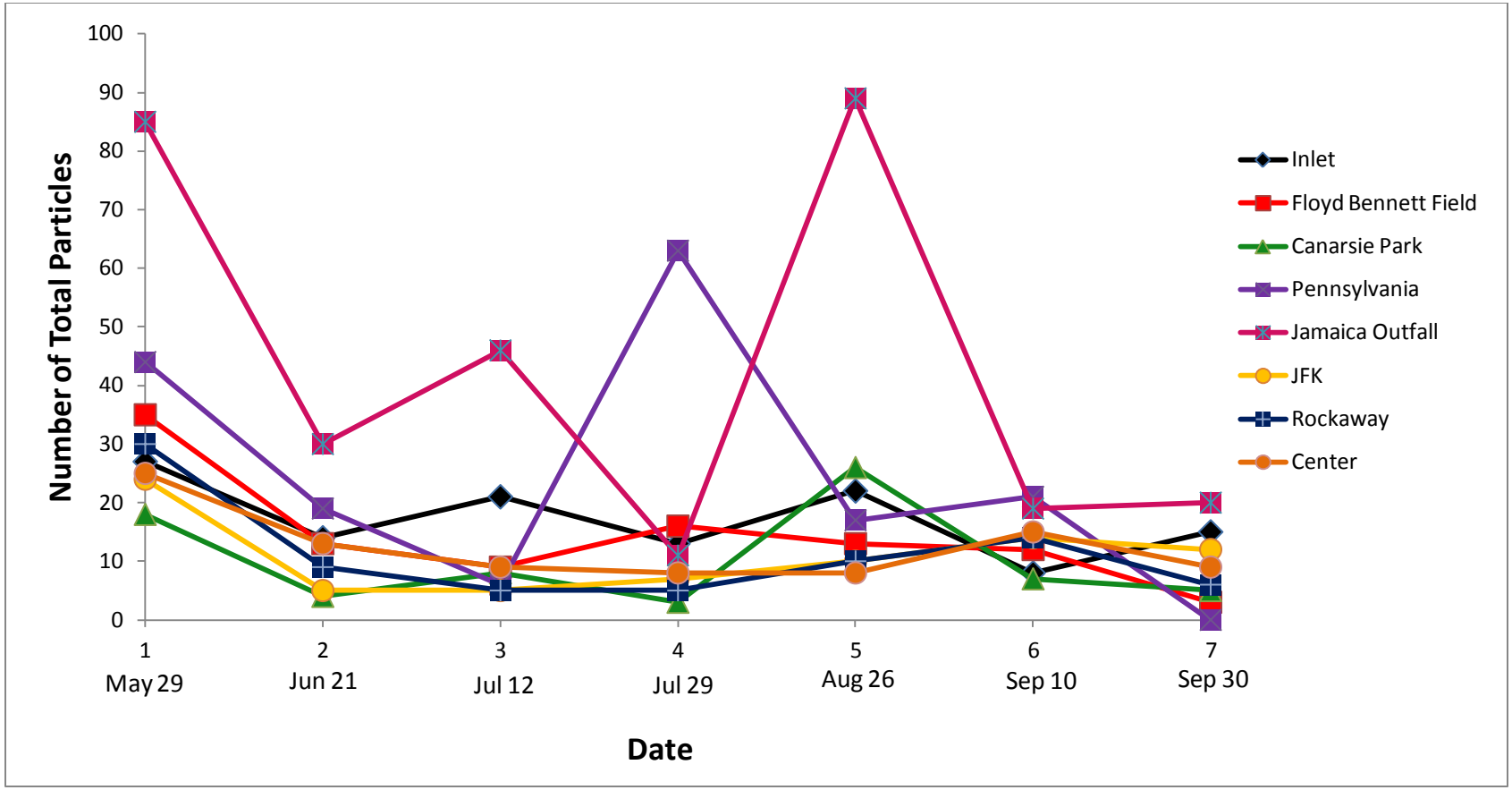


Figure 16A Friedman test results comparing the number of total particles to dates and sites sampled for Jamaica Bay.

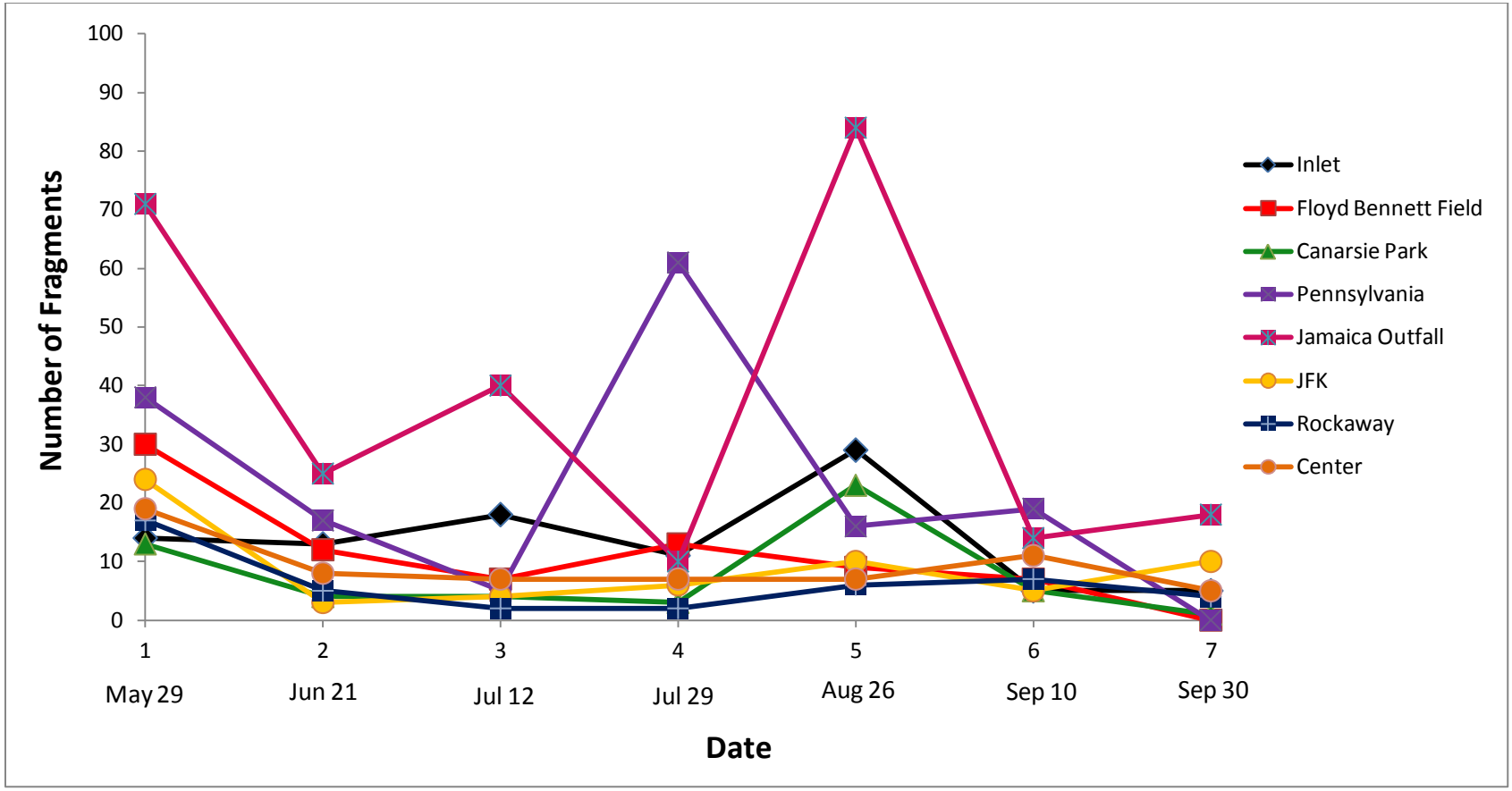


Figure 16B Friedman test results comparing fragments to dates and sites sampled for Jamaica Bay.

Table 1 Concentrations of various POPs found on microplastic particles

POP Compound	Concentration[a] (ng ⁻g)	Location[b]	Source
BPA	0-730	Atlantic and Pacific, coastal and open water	Hirai et al. 2011
DDE	0.16-3	Coastal Japan	Mato et al. 2001
DDTs (21 congeners)	22-7100	Pacific, coastal and open water	Rios et al. 2007
DDTs (DDT, DDE, DDD)	0-4	Coastal Portugal	Frias et al. 2010
DDTs (DDT, DDE, DDD)	0-198	Atlantic and Pacific, coastal and open water	Hirai et al. 2011
NP	0.13-16 μg ⁻g	Coastal Japan	Mato et al. 2001
NP	0-3936	Atlantic and Pacific, coastal and open water	Hirai et al. 2011
PAHs (16 congeners)	39-1200	Pacific, coastal and open water	Rios et al. 2007
PAHs (15 congeners)	0.2-320	Coastal Portugal	Frias et al. 2010
PAHs (15 congeners)	1-9297	Atlantic and Pacific, coastal and open water	Hirai et al. 2011
PAHs (25 congeners)	21-1120	San Diego Bay, CA	Rochman, Manzano et al. 2013
PBDEs (20 congeners)	0.02-9909	Atlantic and Pacific, coastal and open water	Hirai et al. 2011
PCBs (1 congener)	5 ppm	New England coastal waters	Carpenter et al. 1972
PCBs (23 congeners)	4-117	Coastal Japan	Mato et al. 2001
PCBs (36 congeners)	27-980	Pacific, coastal and open water	Rios et al. 2007
PCBs (15 congeners)	0.02-16	Coastal Portugal	Frias et al. 2010
PCBs (39 congeners)	1-436	Atlantic and Pacific, coastal and open water	Hirai et al. 2011

[a]All concentrations in ng ⁻g unless otherwise specified in the table.
[b]"Coastal" indicates beach or nearshore samples, "open water" indicates pelagic surface water samples.

Table 2 Summary of macroplastic found

Location			
Bay	Sample Site	Particle Description	Size (cm)
Jamaica	Center	Green fragment	0.9331
	Center	White fiber	1.2672
	Pennsylvania	Clear film	2.956
	Pennsylvania	Clear fragment (Band-Aid)	2.984
	Pennsylvania	Fragment (food wrapper)	5.307
	Rockaway	White pellet	0.459
	Rockaway	White fragment	1.347
	Rockaway	Clear film	2.742
	Jamaica Outfall	Yellow fragment	2.618
	Jamaica Outfall	Clear film (Zip-lock)	2.809
	Floyd Bennett Field	Clear film	4.998
	Floyd Bennett Field	White film	3.296
Shinnecock	Cormorant Point	Blue fiber	0.5213
	Pine Neck	Red fiber	0.5991
	Inner Tiana Bay	Black fiber	0.6416
	Ponquogue Bridge	Black fiber	1.593
	South Grass	White fiber	3.380
	South Grass	Black fiber	0.5348

Table 3 Summary of Friedman test results

Location	Test Type	Particles	P-value	Degrees of Freedom	Chi-square Value
Jamaica Bay	Spatial, date as block factor	Total	0.0033*	7	21.3362
		Fragments	0.002*	7	22.5475
		Fibers	0.6426	7	5.1421
	Temporal, site as block factor	Total	0.0047*	6	18.6772
		Fragments	0.0025*	6	20.2825
		Fibers	0.1295	6	9.8875
Shinnecock Bay	Spatial, date as block factor	Total	0.4535	10	9.8532
		Fragments	0.3553	10	11.0282
		Fibers	0.6881	10	7.3908
	Temporal, site as block factor	Total	2.173×10^{-5}*	8	35.4934
		Fragments	0.04*	8	16.1782
		Fibers	9.639×10^{-5}*	8	31.9169

* indicates statistical significance.

Table 4 Comparison of global microplastic concentrations

Location	Quantity (items m^{-3})	Source
Portugal, coastal waters	0.036	Frias et al. 2014
Long Island Sound	0.05-0.10	Carpenter et al. 1972
Rhode Island Sound	0-0.10	Carpenter et al. 1972
Shinnecock Bay	**0-0.58**	**This Study**
Block Island Sound	0-2.5	Austin & Stoops-Glas 1977
Northeast Pacific coastal waters	0-3.14	Doyle et al. 2011
Jamaica Bay	**0-3.93**	**This Study**
Western Italy, coastal waters	0-9.67	Fossi et al. 2012
Niantic Bay, CT	0-14	Carpenter et al. 1972
Singapore, coastal waters	0-200	Ng & Obbard 2006
Western Sweden, coastal waters	167-102,550	Noren 2007