Science of the Total Environment 664 (2019) 753–760

Contents lists available at ScienceDirect

Science of the Total Environment

journal homepage: www.elsevier.com/locate/scitotenv

Characterization of microplastics in the surface waters of Kingston Harbour

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HIGHLIGHTS

- Microplastic pollution is evident in surface waters of Kingston Harbour, Jamaica.
- Microplastics concentration ranged from 0 to 5.73 particles/m^3 (0–2,697,674.13 particles/km^2).
- Fragments were the most abundant morphology sampled from Kingston Harbour.
- The average microplastic:zooplankton ratio was 0.18%.

GRAPHICAL ABSTRACT

ARTICLE INFO

Article history:
Received 9 November 2018
Received in revised form 17 January 2019
Accepted 24 January 2019
Available online 05 February 2019

Editor: Jay Gan

Keywords:
Small island developing states
Kingston Harbour
Microplastic
Zooplankton
Jamaica
Caribbean
Marine debris

ABSTRACT

Microplastic contamination of the marine environment has garnered global attention in recent years, and its distribution and effects in many small island developing states (SIDS) are still undetermined. As such, this study serves to detail an investigation of the abundance, spatial distribution and characteristics of surface water microplastics in the Kingston Harbour, a heavily polluted embayment in Jamaica. Fortnightly sampling with a manta trawl (335 μm mesh) revealed non-variable concentrations of 0–5.73 particles/m^3 (0–2,697,674.13 particles/km^2) across stations adjacent to mangrove forests, key nursery grounds for many commercially important finfish and shellfish. Microplastics found in samples were predominantly fragments and were between 1 mm and 2.5 mm. Fourier Transform Infrared (FT-IR) spectroscopy identified polyethylene and polypropylene in fragments selected for analysis. These data serve to establish a crucial baseline of the status of microplastic pollution in Kingston Harbour.

1. Introduction

Since the start of production in the 1950's, an estimated 8.3 billion metric tons (8300×10^6 MT) of plastic have been produced, up to

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https://doi.org/10.1016/j.scitotenv.2019.01.319
0048-9697/

2016 (Geyer et al., 2017). In 2015, annual global plastic production reached 322 million metric tons (PlasticsEurope, 2016) and up to 12.8 million metric tons ended up in the sea (Jambeck et al., 2015). In many countries, the exact quantities of plastic contamination are not known and only fractions of the material are tallied through efforts such as coastal clean-ups and other recovery activities. One European report stated that single-use plastic products constituted an average of 51% of the litter recovered on beaches (Seas at Risk, 2017). Upon this basis, many countries worldwide have devised policies and implemented legislations for the ban of the use and manufacture of these products (Xanthos and Walker, 2017; UNEP CEP, 2018). At the time of writing this manuscript, the government of Jamaica had made significant strides in imposing a ban, effective January 2019, phasing out the importation, manufacture and usage of all Styrofoam products, as well as single use polyethylene shopping bags and plastic straws (Jamaica Gleaner, September 18, 2018). Voluntary efforts in 112 countries from the 2016 International Coastal Clean-up day alone, recovered about 18.4 million pounds of marine debris from roughly 15,000 km of coastline (Ocean Conservancy, 2017); over 80% of which was comprised of plastic (Moore, 2008; Cole et al., 2011).

Marine plastic pollution challenges especially many of the small island developing states (SIDS) on several fronts: their geographical location relative to many of the marine litter hotspots threatens their shores and wildlife; their economies are heavily dependent on activities surrounding the marine environment (tourism, fisheries, trade); their current waste management infrastructure is inadequate to properly contain the country's waste and sanction indiscriminate activities in that regard; their financial resources are indisposed to allocate funding for mitigating marine litter issues (Lachmann et al., 2017).

The dominance of plastics in the marine environment has manifested several threats to marine organisms and their environment. Larger marine animals have often been found as victims of entanglement, choking, malnutrition and premature death because of their encounters with large debris during feeding and locomotion (Green, 2014). In addition to the physical hazard, the hydrophobicity of plastics has been related to the metabolic transfer of many organic and inorganic compounds such as heavy metals, plasticizers and common persistent organic pollutants, that adsorb to the surface of these plastics (Rochman et al., 2013; Rochman et al., 2014).

The unrecovered plastic debris weather overtime and break up into smaller and smaller fragments called microplastics, thus increasing pollutant concentration and potential bioavailability to smaller marine organisms. Microplastics are generally defined as plastic particles that have at least one dimension measuring 5 mm or less (Barnes et al., 2009; Andrady, 2011; Au et al., 2017), and can be further classified according to the purpose for which the material was formed. Microplastic particles either enter the environment as primary microplastics, which were manufactured as tiny microbeads, such as those used in cosmetics; or secondary microplastics, which were fragmented from larger debris due to weathering; by microbial degradation, UV light exposure and physical deterioration through wave action (Cole et al., 2011). At these sizes, plastics in coastal productive waters, which have been shown to bind to microalgae or adhere to the setae of feeding appendages (Lusher et al., 2017), can be mistaken for food and ingested by a range of marine organisms, especially filter feeders as are found in coastal habitats like Kingston Harbour and its mangroves (Cole et al., 2013; Steer et al., 2017).

Kingston Harbour is Jamaica's most contaminated bay, through its proximity to the country's capital city and the harbour's natural physiography (Webber and Webber, 1998), which makes it almost completely enclosed with a relatively small opening at its western end. Kingston Harbour is reputed to be the world's 7th deepest natural harbour with only maintenance dredging being required for most of its navigable areas. The Harbour has been extensively researched, with studies documenting the increasing organic pollution (eutrophication) for over three decades and an entire volume of the Bulletin of Marine Sciences (Webber and Webber, 2003) being dedicated to research conducted in Kingston Harbour. While the most significant contaminants of the harbour have historically been organic in nature, the past ten years have seen a significant increase in the amount of solid waste entering the Harbour via the 19 storm drains (gullies) and two rivers that enter the 51km^2 waterway. In 1993–1994, Green and Webber (1996) assessed solid waste in the mangrove areas of Kingston Harbour, of which plastic debris was found to be the most abundant and frequently encountered material. Green (2014) characterized marine litter on Refuge Cay (an ecologically important mangrove island found in the Kingston Harbour) and the effects of the material on the important bird populations found on the cay. However, both these studies focused on macro-plastic debris (smallest size sampled being 50–150 mm).

While microplastics have been documented in marine environments worldwide, their concentrations and implications have never been assessed in Kingston Harbour or anywhere else in Jamaica. A study was, therefore, designed to investigate quantities and types of microplastic debris in Kingston Harbour with a focus on the mangrove areas, which receive much of the macro-plastic debris brought into the harbour. These areas are also important nursing grounds (Aiken et al., 2009) for commercially important fish and shell-fish that are extracted from the Harbour and south coast shelf of Jamaica.

The study assesses the quantities of microplastics in Kingston Harbour's mangrove areas and compares these quantities to zooplankton which have already been extensively quantified for the water body. Furthermore, this study will provide valuable baseline information about microplastic contamination, in light of the legislation to reduce the volume of single-use plastics in Jamaica.

Our study questions were therefore as follows:

1. Are there measurable quantities of microplastics in the Kingston Harbour and its mangrove areas?
2. What are the quantities of microplastic debris and how do these abundances compare to zooplankton abundances in the water body?
3. What are the types and size ranges of microplastic debris and can these facilitate identification of origin/sources of the material?

2. Materials and methods

2.1. Sample collection

Sampling was conducted fortnightly (September 14, November 2, November 16, December 1 and December 15, 2017) at four stations (Fig. 1). Stations included; a control (Ctrl.), located outside the Harbour adjacent to the town of Port Royal; stations within the harbour adjacent to Gallow's Point (GP); Refuge Cay (RC); Buccaneer beach (BB), the latter at the eastern end of the Harbour. Stations inside the harbour were selected near the southern shore along the Palisadoes tombolo, which is home to many species of juvenile fish and shellfish and is threatened by the accumulation of debris that constantly drifts south across the harbour from the gullies and rivers. All microplastics samples ($n = 40$) were collected in duplicate trawls at each station between 10 a.m. and 12 noon using a 335 μ mesh manta trawl (Fig. 2) (obtained from 5 Gyres Institute as part of the Trawl-Share programme). The trawl had a 24″ × 9.84″ opening and was towed along a transect at each station for 15 min at a speed of ~1 knot and ensuring that the net sampled outside of the wake of the boat (Lippiatt et al., 2013). At the end of each trawl, the contents of the 335 μ cod end bag were transferred into clean (acid-washed with 10% HNO_3 and rinsed thoroughly with deionized water (18.2 MΩ)), labelled glass jars using 500 mL of water from the respective station and the bag rinsed thoroughly with seawater. The bottles were placed in a cooler with ice packs for preservation and brought to the laboratory for analysis. Zooplankton collections were done using a 335 μ plankton net with a 0.5 m hoop diameter which was towed for 5 min just below the surface. The contents of the cod end were

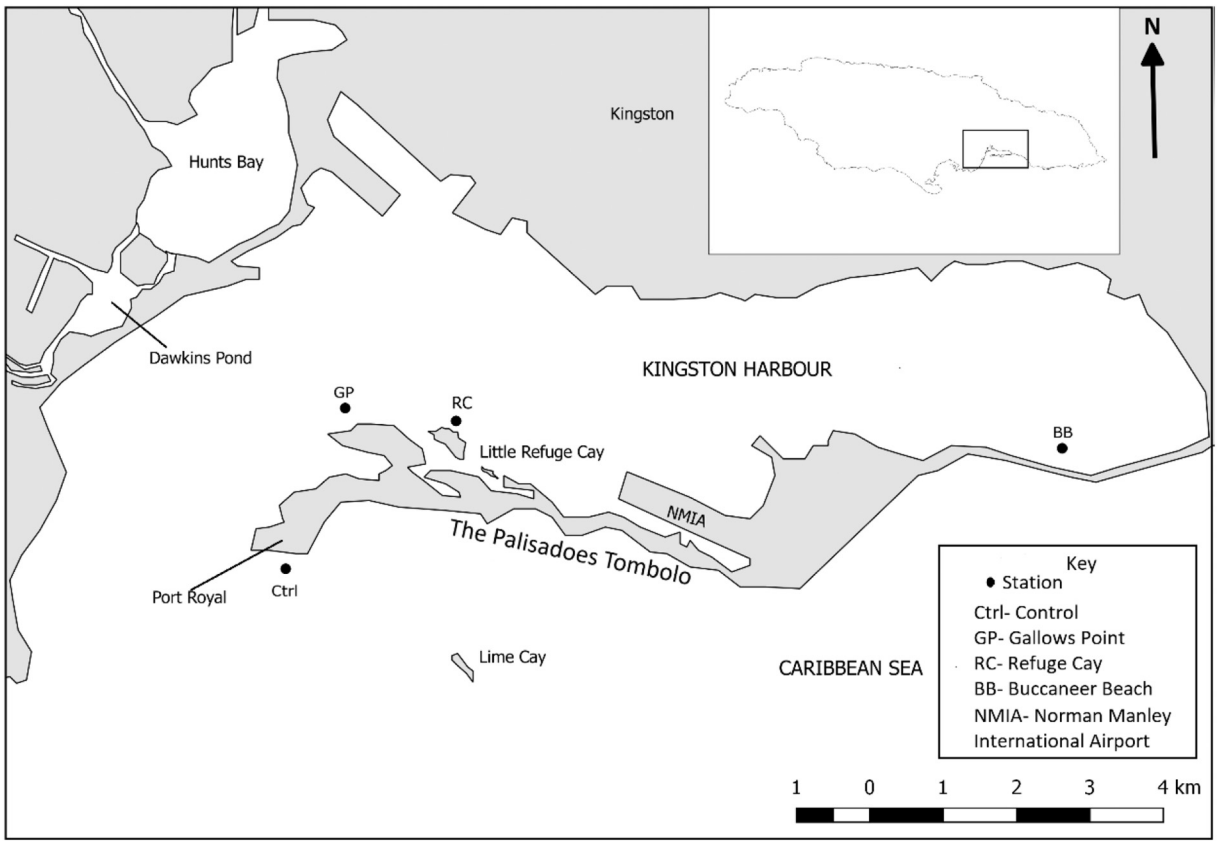

Fig. 1. Google image of Kingston Harbour and the Palisadoes tombolo showing the location of the four stations sampled for zooplankton and microplastics.

transferred to labelled bottles containing 10 mL of 200 proof ethyl alcohol for immediate preservation prior to enumeration in the laboratory. Flow meters (General Oceanics L6) were placed across the mouth of both nets to record the length of tow and hence the volume of sea water sampled. In addition, GPS Coordinates were recorded at the beginning and end of each tow using a handheld GPS (Garmin GPSMAP 62).

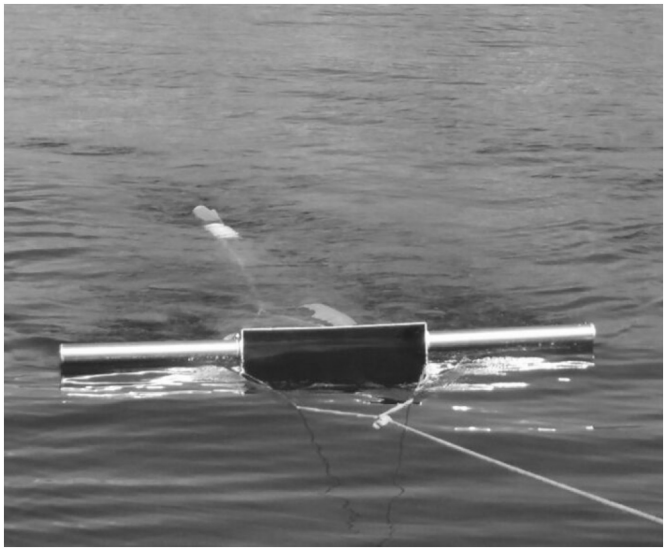

Fig. 2. The manta trawl (obtained from 5 Gyres institute) used in sampling surface waters in Kingston Harbour for microplastics.

2.2. Laboratory analysis

2.2.1. Microplastics

2.2.1.1. Wet sieving, digestion and density separation. Microplastics samples were prepared for analysis using modifications of the protocol outlined by Masura et al. (2015). Samples were wet sieved through 5 mm and 0.25 mm stacked sieves. Material retained on the 0.25 mm (250 μm) sieve was transferred to a beaker and placed in an oven at 90 °C for 24 h or more until completely dry. The organic material was oxidized with 20 mL aliquots of 0.05 M Fe(II) solution and 30% H_2O_2 on a hot plate at 75 °C for 30 min, with additional 20 mL aliquots of 30% H_2O_2 added every 30 min until oxidation was complete. Lusher et al. (2017) reported 95% recovery rates and potentially 6.2% loss in size for PE and PP polymers using a comparable method. A saturated NaCl solution (~5 M) was used to facilitate the separation of the plastics from heavier non-polymeric materials by the addition of 6 g of NaCl per 20 mL of peroxide. The suspension was covered with foil and allowed to separate overnight in the density separator funnel. Floating plastics were then collected on a 0.25 mm (250 μm) sieve and rinsed free of the hypersaline solution with distilled water. The settled portion was also passed through the sieve and assessed for any additional microplastics. Collectively, the microplastic particles were dried and were placed in a vial prior to microscope separation, counting and classification.

2.2.1.2. Quality assurance and contamination control. Contamination control procedures were employed, with utmost care, to minimise the chance of plastic particles being introduced to the samples during sample collection, preparation and laboratory analysis. Glass jars (cleaned with dilute HNO_3 and rinsed thoroughly with 18.2MΩ deionized water) were used to containerize samples collected for further

processing in the laboratory. Glassware (beakers, watch glasses, funnels) and stainless-steel apparatus (sieves, forceps, spatulas) were used during sample processing and aluminium foil used to cover samples to minimise contamination from airborne fibres. A laboratory coat and nitrile gloves were always worn while work was carried out in an enclosed laboratory away from foot traffic.

To avoid misidentification of microplastics using a microscope, a particle was established as microplastic based on visual criteria described by Nor and Obbard (2014) and Löder and Gerdts (2015), ensuring that there are no visible cellular or organic structures, coloured particles are homogenous in colour, transparent particles are viewed under high magnification to exclude a biological origin and fibres should be equally thick with three-dimensional bending to exclude a biological origin. Microscopic examination was carried out in a laboratory designed with sealed windows and passage through the lab was restricted to minimise airborne contamination. Procedural blanks using deionized water were passed through the entire analytical process and treated in the same manner as were samples, to identify any possible points of contamination. These tested negatively for contamination.

2.2.1.3. Visual identification and classification. Prepared samples were illuminated with gooseneck lighting (Schott ACE 1) and viewed under a Meiji EMZ8TR stereomicroscope at a magnification of ×40. Microplastics were separated from any apparent undigested organic material and categorized according to shape (fragment, fibre, foam, bead) and size (0.335–1 mm, 1–2.5 mm. 2.5–5 mm, >5 mm) (Zhao et al., 2014). Samples were photographed at a magnification of ×7 with the aid of Lumenera Infinity Analyze software. Microplastics abundance in particles/km^2 and particles/m^3 was then computed from the tally and the distanced towed and the dimension of the opening of the manta net.

2.2.1.4. FT-IR analysis. Following visual sorting and classification, Fourier Transform Infrared (FT-IR) spectroscopy was employed to identify polymer composition of the microplastic particles using a Bruker Vector 22 FT-IR spectrometer equipped with a deuterated triglycine sulphate (DTGS) detector, Zinc Selenide crystal and clamp. Fragments were analysed from samples taken from each of the four (4) stations for analysis because of their high visibility to the naked eye and ease of transfer to and from the crystal. The crystal was cleaned with lint-free paper and methanol and a background scan performed before each particle was analysed. Particles were analysed in transmission mode at a speed of 5 Hz, within the range of 4000–600 cm^{-1} and a combination of 40 scans per analysis. The resulting spectra were processed in the accompanying Opus 65 software and were compared with the pristine FTIR spectra of common polymers (polyethylene, polystyrene, polypropylene, nylon, polyvinyl chloride) obtained from the BIORAD Spectrabase spectral library.

2.2.2. Zooplankton

Zooplankton collections which were stored in 90% ethanol were homogenized and split into requisite sub-samples (Van Guelphen et al., 1982) before a tally of zooplankton was determined by counting totals from sub-samples poured into a Bogorov counting tray and viewed under a Wild dissecting microscope (Mag ×60). Zooplankton were enumerated based on major taxonomic groups; Medusae, Cladocera, Copepoda, Decapoda, Chaetognatha, Larvacea, Ichthyoplankton and 'other'. Total numbers were converted to numbers per m^3 based on volume of water sampled obtained from the flow meter readings (Francis et al., 2014; Lue and Webber, 2014; Webber et al., 2015).

2.3. Statistical analysis

All data were tabulated into spreadsheets in Microsoft Excel 2010 and statistical manipulations performed using XLSTAT (2008). The Sharipo-Wilks and Lilliefors tests were used to determine normality and the data were subsequently log transformed for homogenization. One-way ANOVA was applied to determine significance in spatial and temporal variability for zooplankton abundances and microplastics concentrations across stations, followed by a Tukey's honestly significant difference (HSD) post hoc test and Dunnett's test to assess similarities between stations. All tests were performed at a 95% confidence level.

3. Results

Overall, the results confirmed surface water microplastic contamination at all four stations sampled on the 5 days of this study period ($n = 1702$). The mean microplastic concentration for the area over the sampling period was 0.76 particles/m^3 (359,593.41 particles/km^2). Generally, concentrations differed greatly throughout the study period (Fig. 3). On one occasion at RC (Refuge Cay), no microplastics (0 particles/m^3; 0 particles/km^2) were sampled. In another instance, the highest microplastic concentration (5.73 particles/m^3; 2,697,674.13 particles/km^2), was recorded on a single occasion at BB (Buccaneer Beach). A one-way ANOVA confirmed significant temporal variability ($p = 0.014$) in microplastic concentrations during the study period. There was, however, no significant spatial variability of microplastic concentrations across stations ($p = 0.519$). Pair-wise comparisons using Tukey's HSD test and Dunnett's test for comparison with the control (Ctrl) station further revealed microplastic tehomogeneity across stations.

Zooplankton abundances were dominated by the Copepoda (71%) followed by Crustacea larvae (16%), the decapod *Lucifer faxoni* (5%) and the cladoceran *Penilia avirostris* (4%). Zooplankton abundance at each station (Fig. 4) was separated (by Tukey's multiple range test) into three discrete station groupings, in like manner to microplastic concentrations, with Stations GB and GP in one group and Stations Ctrl and RC segregated into two separate groups.

Furthermore, there was significant variability in the spatial distribution of zooplankton abundance across stations (ANOVA, $p \leq 0.001$), with Refuge Cay (RC) having the highest mean abundances overall (1.92×10^3–1.32×10^4 individuals/m^3). On the contrary, zooplankton abundances exhibited no significant temporal variability (ANOVA, $p = 0.409$), across sampling dates. Mean zooplankton abundances were generally lowest at Ctrl, ranging from 0.12×10^3–0.30×10^3 individuals/m^3 throughout the study period. The ratios of microplastic abundance (n/m^3) to zooplankton abundance (n/m^3) counts were used to estimate encounter probabilities for planktivores. Microplastics: zooplankton ratios averaged 0.18% and were generally low overall, with the highest ratio (1.74%) observed at the Control (Ctrl) station on September 14, 2017.

Twenty four percent (24%) of the microplastics sampled were estimated to be between 0.335 mm and 1 mm, having similar dimensions to that of common species of zooplankton (*Acartia tonsa*, *Penilia avirostris*, *Temora turbinata*, *Paracalanus* spp.), which have been used as bioindicators of pollution in the Kingston Harbour in the past (Francis et al., 2014). However, majority of microplastics were classified (Fig. 5) between 1 mm and 2.5 mm (47%), while others were between 2.5 mm and 5 mm (22%) and fewer having a dimension >5 mm (7%).

Microscopic examination of the recovered plastics also facilitated the categorization of the particles into four main types: fragment, fibre, foam and microbead (Fig. 6). Fragments accounted for 86.08%, with fibres second in abundance (12.68%) followed by foam (0.92%) and beads (0.31%). Microplastics were also identified by a wide range of colours, with most being transparent (35%), opaque (27%), white (19%) and black (10%). Others were either blue (5%), green (2%), red (1%), yellow (1%) or multi-coloured (1%).

From the particles enumerated, a representative group of fragments were selected for FT-IR analysis from samples collected at the 4 stations from the 5 sampling occasions. Spectra generated from the analysis of fragments were compared with pristine spectra of the common polymers, polyethyelene, polypropylene and polystyrene from BIORAD's

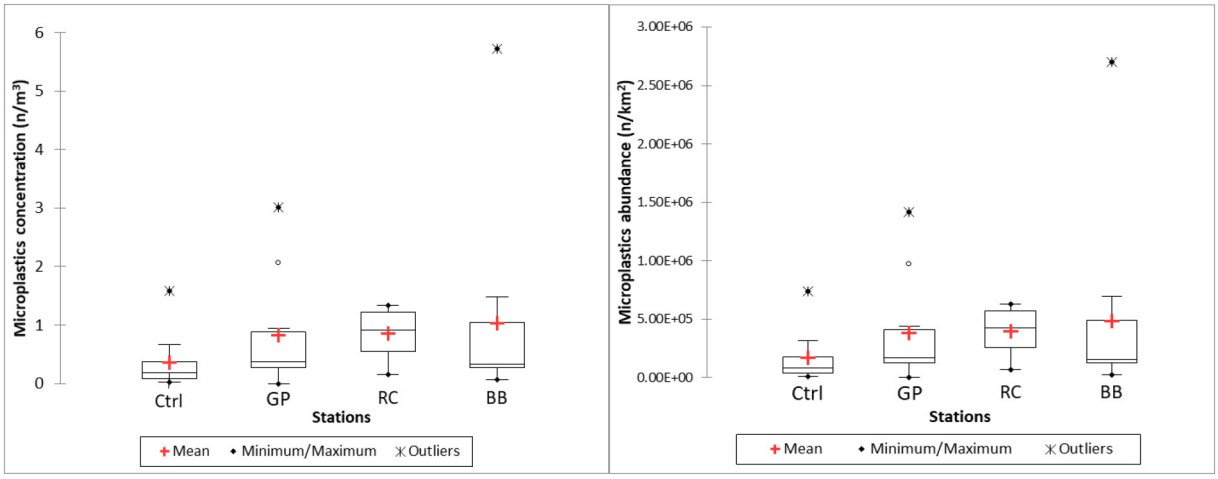

Fig. 3. Microplastic abundance in n/m^3 (*Left*) and microplastic abundance in n/km^2 (*Right*) of the four stations sampled between September and December 2017. Control (Ctrl) station just outside tombolo; Gallows Point (GP); Refuge Cay (RC); Buccaneer Beach (BB).

Spectrabase online comparison tool. Polyethylene (78%) dominated the fragments analysed while polypropylene was found in 22% of the fragments analysed. However, the application of FT-IR in this study revealed that there were non-polymer particles that were initially enumerated as microplastic (Fig. 7) and has re-emphasized the importance of spectroscopic analysis for polymer identification in microplastics studies.

4. Discussion

These findings indicate the presence of measurable quantities of microplastics in the surface waters of Kingston Harbour. Though several studies have been published on plastic and microplastic debris in relation to the Caribbean and by extension, small island developing states (Lachmann et al., 2017; Bosker et al., 2018), limited knowledge exists for surface water microplastics concentrations in surface waters of these regions. However, several other reports of surface water microplastics concentrations exist in other regions (Aytan et al., 2016; Collignon et al., 2012; Di Mauro et al., 2017; Gündoğdu, 2017; Lattin et al., 2004; Zhao et al., 2014). The mean microplastic abundance reported in this study (0.76 particles/m^3) were similar that found in the Bohai Sea, China (0.33 particles/m^3, Zhang et al., 2017), but appreciably different from other studies in the Yangtze estuary, China (4137.3 particles/m^3; Zhao et al., 2014), the SE Black Sea (600–1200 particles/m^3; Aytan et al., 2016) and on the opposite end of the spectrum, the west coastal waters of Sweden (0.01–0.04 particles/m^3; Norén, 2007). A study reporting concentrations in particles/km^2 by Gündoğdu (2017) in the Iskenderun Bay, Turkey (mean of 1,067,120. particles/km^2) was roughly 10-fold higher than in the Kingston Harbour (359,593.41 particles/km^2).

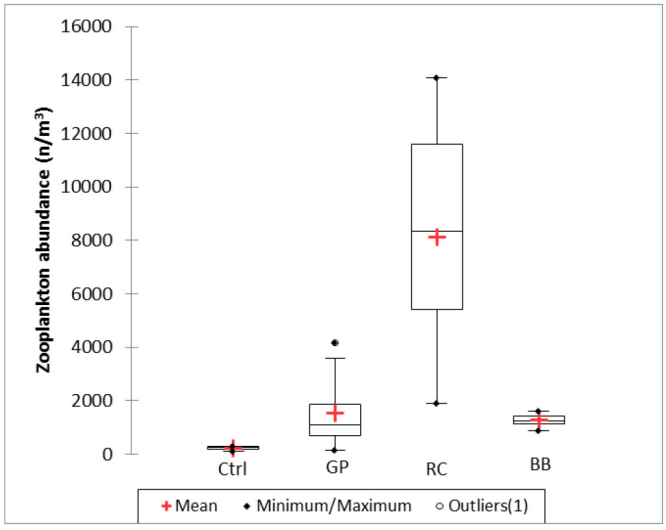

Fig. 4. Total zooplankton abundances collected between September and December 2017 at the four stations in the study area of Kingston Harbour. Control (Ctrl) station just outside of the tombolo; Buccaneer beach (BB); Refuge Cay (RC); Gallows Point (GP).

Each study area is unique to marine debris load, hydrodynamics influencing vertical mixing and the level of aquatic primary productivity, and the efficiency of the sampling equipment used can also affect the concentrations of microplastics estimated (Zhao et al., 2014; Aytan et al., 2016; Song et al., 2018). One study conducted in False Bay, South Africa that evaluated the rate of fouling of polyethylene (HDPE & LDPE) sheets of varying thicknesses (0.1 mm, 0.2 mm, 0.5 mm, 1 mm, 4 mm, 5 mm, 9 mm and 50 mm), tethered 10 cm below the water surface, found that most microplastic fragments were negatively buoyant six weeks after their deployment (Fazey and Ryan, 2016). Field and laboratory experiments done by Kaiser et al. (2017) also highlighted that after 6 weeks of incubation in estuarine and marine waters both polyethylene and polystyrene pellets displayed an increased sinking velocity.

With a history of highly eutrophic waters, zooplankton communities in the Kingston Harbour have been used as bioindicators of the level of organic pollution that has been persistent over the years (Webber and Webber, 1998; Francis et al., 2014). This was reflected in the generally low microplastic:zooplankton ratios throughout the study (Table 1), suggesting that planktivores would have a probability of 0.01–1.74% of encountering a microplastic particle instead of zooplankton. Research to date has noted several consequences of ingestion of microplastics by plankton and planktivorous organisms and continues to be of significant concern (Cole et al., 2013; Lusher et al., 2013; Rochman et al., 2014; Desforges et al., 2015). Various bacterial colonies and microorganisms residing on the surface of these plastics may have contributed to biofouling of surface water microplastics, causing increased density and eventual submersion (Kaiser et al., 2017). Therefore, surface water microplastics in Kingston Harbour may have been underestimated, and additional work would be pivotal to assessing microplastic distribution throughout the water column.

The widely used wet peroxide oxidation (WPO) protocol by Masura et al., 2015 employed in this study successfully preconcentrated the microplastics present in the samples, but possibly could have partially or completely dissolved some particles (<1 mm), resulting in an underestimation of initial particle size (before digestion) and overall tally (Lusher et al., 2017; Miller et al., 2017). Factors such as degree of

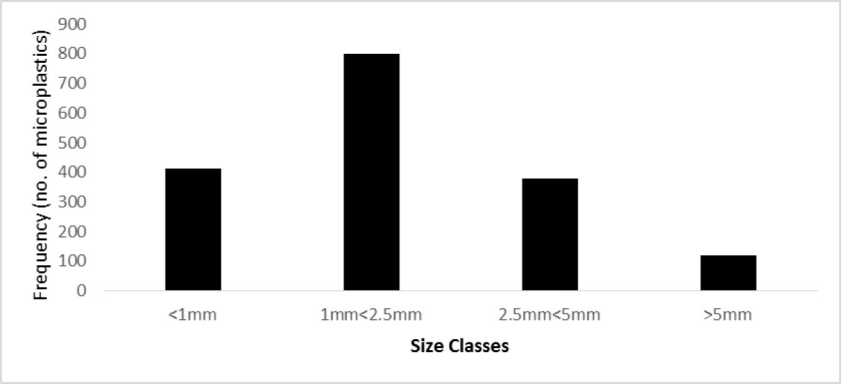

Fig. 5. Size classification of microplastics sampled in the Kingston Harbour between September and December 2017. Bars indicate frequency in each size class.

weathering of the microplastics, size, shape, peroxide concentration, temperature and exposure time to oxidation may impact the degree to which the microplastic particles are impacted by the WPO process (Gewert et al., 2017; Lusher et al., 2017; Munno et al., 2018). Other proposed methods using acidic and alkaline reagents for digestion have also been found to destroy the microplastics in the samples (Cole et al., 2014; Enders et al., 2017). On the contrary, enzymatic digestion has shown to have negligible impact on the microplastics retrieved from biogenic-rich samples (Cole et al., 2014; Löder et al., 2017) but remains a costly option for microplastic preconcentration compared to the latter-mentioned alternatives.

Microplastics sampled in this study were varied in size but were mostly (47%) between 1 mm and 2.5 mm, 24% between 0.335 mm and 1 mm, and 22% between 2.5 mm and 5 mm. Stations at Gallows Point and Refuge Cay recorded the highest number of microplastics enumerated between 0.335 mm and 5 mm, and are notable for being two of the most severely polluted mangrove forested areas in the Kingston Harbour by large anthropogenic debris. At sizes <1 mm, they are bioavailable to a wider range of species of plankton and other filter feeders such as oysters. Additionally, planktivores would also be susceptible to accidental ingestion at these sizes.

Fragments were the most frequently encountered morphology (86.04%) and is an indication that microplastic contamination in Kingston Harbour is predominantly from secondary sources, i.e., from the weathering and fragmentation of meso- and macroplastics (Cole et al., 2011). The high variability in the common shape found in surface waters from previous literature (Nor and Obbard, 2014; Zhao et al., 2014; Gewert et al., 2017; Gündoğdu, 2017) is unique to the main anthropogenic influence (effluents from wastewater treatment plants, typical consumer products used near that region, waste management infrastructure) that contributes to microplastic pollution of the study area.

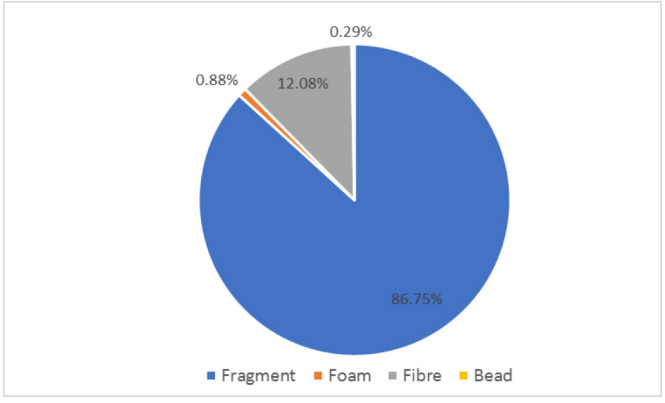

Fig. 6. Morphologies of microplastics sampled in the Kingston Harbour between September and December 2017.

The application of FT-IR spectroscopy in the elucidation of polymer type of microplastics sampled, proved an important step in the classification of microplastics found in the Kingston Harbour. Polyethylene and polypropylene were dominant polymer types found and are two of the greatest subsets of plastics manufactured worldwide (Geyer et al., 2017). However, the analysis of certain particles revealed non-polymer spectra generated from FT-IR spectroscopy and highlights the inadequacies of visual identification in confirming microplastics (Hidalgo-Ruz et al., 2012; Löder and Gerdts, 2015; Li et al., 2018). FT-IR spectroscopy (and other spectroscopic techniques such as Raman spectroscopy) is, therefore, a pivotal supplementary forensic tool in microplastic pollution studies and highlights the importance of the appeal for established standard operating procedures in studying and reporting on microplastics.

Notably, microplastic contamination was observed at the control station, which was outside the Harbour and shielded from direct plastic inputs from the northern shore of the Harbour. Neither would this station be influenced by surface outflow currents, based on the study by Webber et al. (2003). Therefore, microplastic concentrations are more likely influenced by currents of the Caribbean Sea transporting debris from adjacent bays or neighbouring land masses and possible rafting on vectors such as *Sargassum*, which was abundant on some sampling days.

5. Conclusion

Microplastic pollution was ubiquitous at all stations in the Kingston Harbour, ranging from 0 to 5.73 particles/m^3 (0–2,697,674.13 particles/km^2). Though quite variable temporally, there was no apparent spatial variability in microplastic distribution, but the potential threat to marine life that spawn in this region is noteworthy. Surface water microplastic pollution in Kingston Harbour is mainly

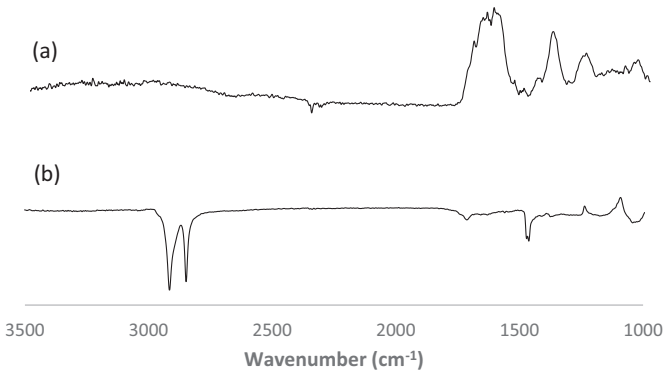

Fig. 7. FTIR spectra of a (a) non-polymer particle initially enumerated in microscopic visual assessment and (b) polyethylene-based particle sampled from Kingston harbour.

Table 1
Microplastics and zooplankton abundances at the four stations of this study; Control (Ctrl), Gallows Point (GP), Refuge Cay (RC), Buccaneer Beach (BB).

Station	Replicates	Date (dd/mm/yyyy)	Microplastics abundance	Microplastics concentration (n/km^2)	Microplastics concentration (n/m^3)	Zooplankton abundance (n/m^3)	MP:Zooplankton ratio (%)[a]
Ctrl	1	14/9/2017	17	314165.96	0.67	147	0.45
Ctrl	2	14/9/2017	49	743027.58	1.58	91	1.73
GP	1	14/9/2017	12	192608.87	0.41	862	0.05
GP	2	14/9/2017	81	1416302.41	3.01	1001	0.30
RC	1	14/9/2017	37	573255.59	1.22	12277	0.01
RC	2	14/9/2017	40	629485.48	1.34	14092	0.01
BB	1	14/9/2017	35	585884.48	1.24	1503	0.08
BB	2	14/9/2017	39	696854.58	1.48	1594	0.09
Ctrl	1	2/11/17	15	82583.30	0.18	308	0.06
Ctrl	2	2/11/17	7	36363.21	0.08	295	0.03
GP	1	2/11/17	33	143625.40	0.30	163	0.19
GP	2	2/11/17	0	0.00	0.00	144	0.00
RC	1	2/11/17	118	561310.54	1.19	1891	0.06
RC	2	2/11/17	68	340934.83	0.72	1951	0.04
BB	1	2/11/17	28	144400.60	0.31	1127	0.03
BB	2	2/11/17	24	123813.12	0.26	864	0.03
Ctrl	1	16/11/2017	38	191544.39	0.41	295	0.14
Ctrl	2	16/11/2017	10	50088.88	0.11	268	0.04
GP	1	16/11/2017	109	441611.33	0.94	3591	0.03
GP	2	16/11/2017	68	972682.00	2.07	4172	0.05
RC	1	16/11/2017	128	630045.03	1.34	6588	0.02
RC	2	16/11/2017	95	517196.76	1.10	6574	0.02
BB	1	16/11/2017	102	2697674.13	5.73	1335	0.43
BB	2	16/11/2017	30	151059.65	0.32	1408	0.02
Ctrl	1	15/12/17	6	28675.38	0.06	201	0.03
Ctrl	2	15/12/17	2	9865.59	0.02	228	0.01
GP	1	15/12/17	64	325324.45	0.69	1217	0.06
GP	2	15/12/17	15	88063.31	0.19	1272	0.01
RC	1	15/12/17	39	243509.74	0.52	10079	0.01
RC	2	15/12/17	52	292176.39	0.62	11378	0.01
BB	1	15/12/17	43	216782.84	0.46	1112	0.04
BB	2	15/12/17	28	155341.41	0.33	1166	0.03

[a] The microplastics:zooplankton ratio is calculated as (number of microplastics/zooplankton abundance) * 100.

attributed to fragmentation of larger plastic debris via mechanical action during their passage to the harbour or persistent wave action as they move about throughout the water column and in the root networks of the mangroves. Polyethylene and polypropylene were the dominant polymer types sampled. Our results also reflect a scenario in a SIDS, where inadequate waste management infrastructure and capital for mitigation efforts for larger plastic debris result in the generation of surface water microplastics. Moreover, these microplastics threaten the longevity of the existing mangrove ecosystem. The importance of this area as a source of fin-fish and shell fish for adjacent communities makes the potential for trophic transfer and human consumption of microplastics very great. Future research must also assess microplastic contamination in organisms found in the Kingston Harbour (water column and benthos).

Acknowledgements

The work was supported by the International Centre for Environmental and Nuclear Sciences (ICENS), the Department of Life Sciences, Centre for Marine Sciences and a GraceKennedy Foundation, James S. Moss Solomon Snr. Grant to M. Webber. The authors wish to thank the staff of the Port Royal Marine Laboratory, University of the West Indies, Jhénelle Williams for giving invaluable assistance in sampling and Raylee Dunkley for his vital contribution to spectroscopic analysis. Great appreciation is also extended to the 5 Gyres Institute for their loan of the manta trawl as part of their Trawl Share programme.

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