Microplastics contamination in Queen Charlotte Sound / Tōtaranui marine sediments

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Executive Summary

There is potent global concern about the presence and consequences of plastic pollution in our oceans. However, many questions remain concerning the origin and fate of micro-sized plastic particles, where they accumulate on and below the seafloor and their effect on the environment, the benthic organisms (e.g. filter-feeders) and up the food chain.

In Aotearoa/New Zealand microplastic particles have successfully been identified in sediments on land and rivers, and in coastal and intertidal zones; but not yet within offshore marine sediments - the ultimate sink for plastic particles. To understand and mitigate the impact of plastic pollution in the ocean it is essential to measure and characterise micro-sized particles in marine sediments and identify their potential sources.

We present the outcomes of the MBIE Envirolink research project (M2140-MLDC160) conducted in the Queen Charlotte Sounds / Tōtaranui (QCS) area. We compare the concentration of plastics contained within sediment samples collected from two seemingly distinct locations in terms of human impact: 1) the high-human impact region of Picton Anchorage; and 2) the near-pristine Kokomohua Marine Reserve.

Using QCS as a pilot study, the University of Auckland and the Marlborough District Council (MDC) worked in partnership with the National Institute of Water and Atmospheric Research (NIWA) and Te Ātiawa o Te Waka-a-Māui iwi Trust Marlborough Sounds (herein referred to as Te Ātiawa Trust). Working with Te Ātiawa Trust was key to better integrate Māori principles and perspectives in responses to environmental change related to human activities.

In this project, the plastic particles from both locations were isolated and quantified to evaluate the accumulation and spatial distribution of plastic contamination / pollutants related to human activities (e.g., runoff, fisheries, marine farms, etc.). Our preliminary results provide the baseline for understanding how human activities on land, coast and in the ocean, are affecting the marine environment of Aotearoa / New Zealand.

The outcomes of this study deliver new data, fundamental to inform environmental policies to mitigate plastic pollution. Having access to information about the source and fate of microplastic particles will directly influence plans for the reduction of plastic pollution entering the coastal marine areas in Marlborough Sounds. This project is the first step towards determining the distribution and impact of microplastic in Aotearoa/New Zealand's marine environment.

Introduction

Unlike larger plastic debris, microplastic particles (< 5 mm) are not readily visible to the naked eye, and relatively little is known about their distribution and accumulation on the seafloor or their impact on the benthic communities. Microplastics can enter the sea in many ways, including through sewage and wastewater systems, riverine inputs, aquaculture and fishing activities. Once in the ocean, microplastic particles can become highly concentrated in specific environments with potentially negative consequences, such as the contamination of marine reserves and negative impacts on marine species and habitats. For example, filter-feeders such as mussels, are highly vulnerable to the ingestion of microplastics, which can then be transferred along the food chain with potential risks for human health). This report will document findings and provide scientific advice to Marlborough District Council (MDC) on the distribution of microplastic particles in marine sediments of Queen Charlotte Sound/Tōtaranui (QCS).

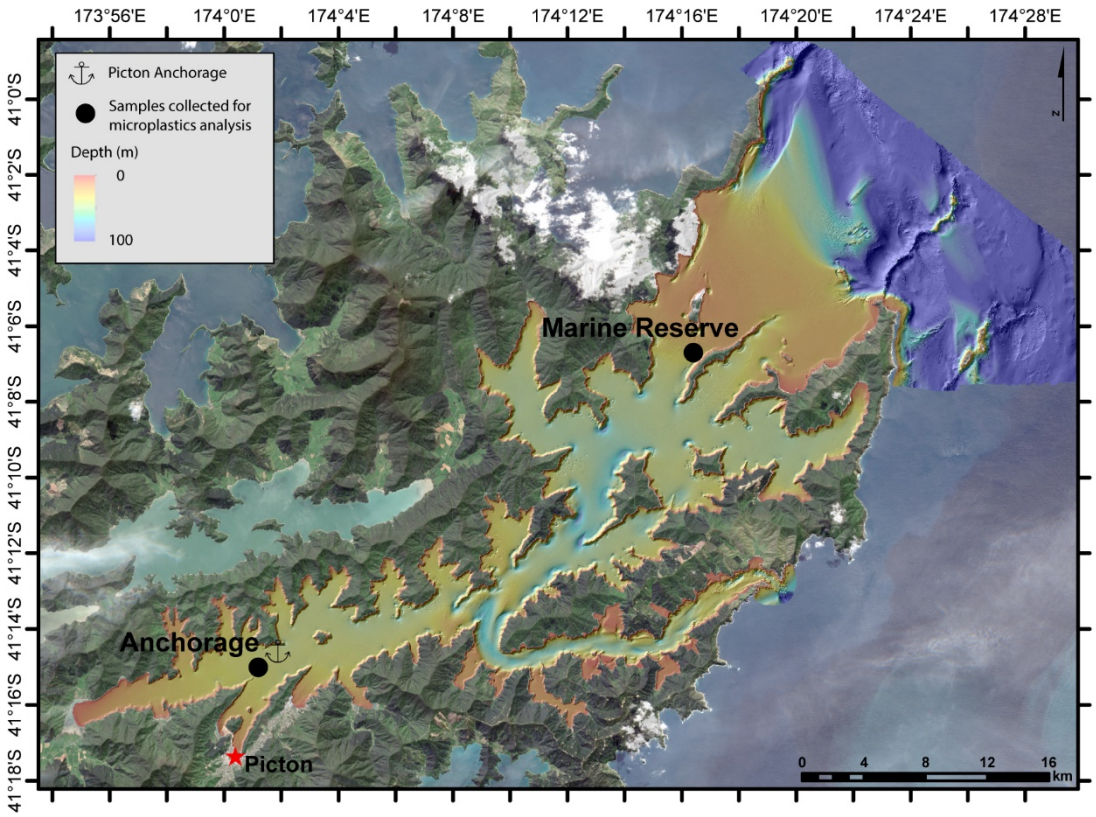

Figure 1. Satellite (LINZ Data Service) and bathymetric (HS51) map of QCS, indicating sites where sediment cores were collected and those that were subsequently analysed for microplastics contamination.

Data collection

In mid-July 2020, a team of scientists including Drs Marta Ribó, Sally Watson and Lorna Strachan embarked upon a three-day marine geology voyage to the QCS. Their goal was to collect sediment cores to investigate human impacts on the shallow marine environment in QCS. The team collected nine sediment cores (a combination of Gravity cores and push cores from a Multi-corer) using a specialised non-plastic Cellulose Acetate Butyrate (CAB) core liners to analyse sediments from the inner and outer QCS for microplastics contamination (Figure 1).

Two coring methods were used:

- KC Denmark Multicorer (Figure 2a) was used to obtain short (<60 cm) cores from the sediment water interface. The coring mechanism is designed to keep the surface intact to enable studies of the upper sedimentary surface layers.
- A Gravity corer (Figure 2b) was used to obtain long (up to 2m) cores. Due to the gravitational component required to obtain these longer cores, the sediment in the upper ~10cm is often destroyed using gravity corers. Gravity cores are typically used to gain a longer cross section of the sediments below the surface. Long cores enable us to collect sediments from before and after human impacts on the marine environment may have started and to measure rates of sedimentation over longer timescales.

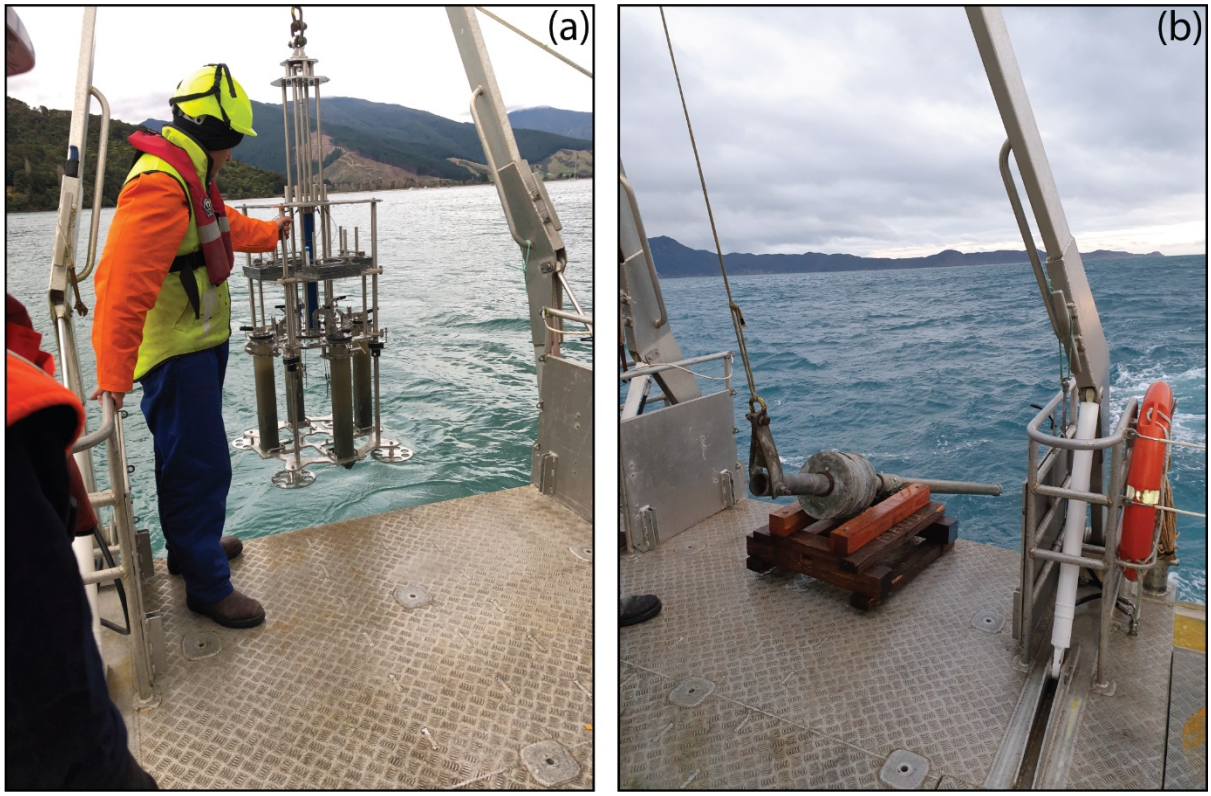

*Figure 2. Pictures of **a)** KC Denmark multicorer and **b)** gravity corer used for collecting core samples.*

Methods

1. Sample processing

Opening cores and sub-sampling

All sediment cores were carefully opened using a radial saw, making sure no CAB debris contaminated the sediment sample (Figure 3). Once the core tube was split in two lengthways, the sample was sliced every 2.5cm for the first 10 cm (sample volume of 88 cm^3), and every 5 cm from 10 cm down to the end (sample volume of 176 cm^3), using metal slicers (Figure 3).

Each sub-sample was stored into a glass beaker (previously cleaned and rinsed with filtered tap water) and covered with clean aluminium foil (Figure 3). Samples with large mass (>100 gr wet weight (w.w.)) were divided and two separate extractions were carried out.

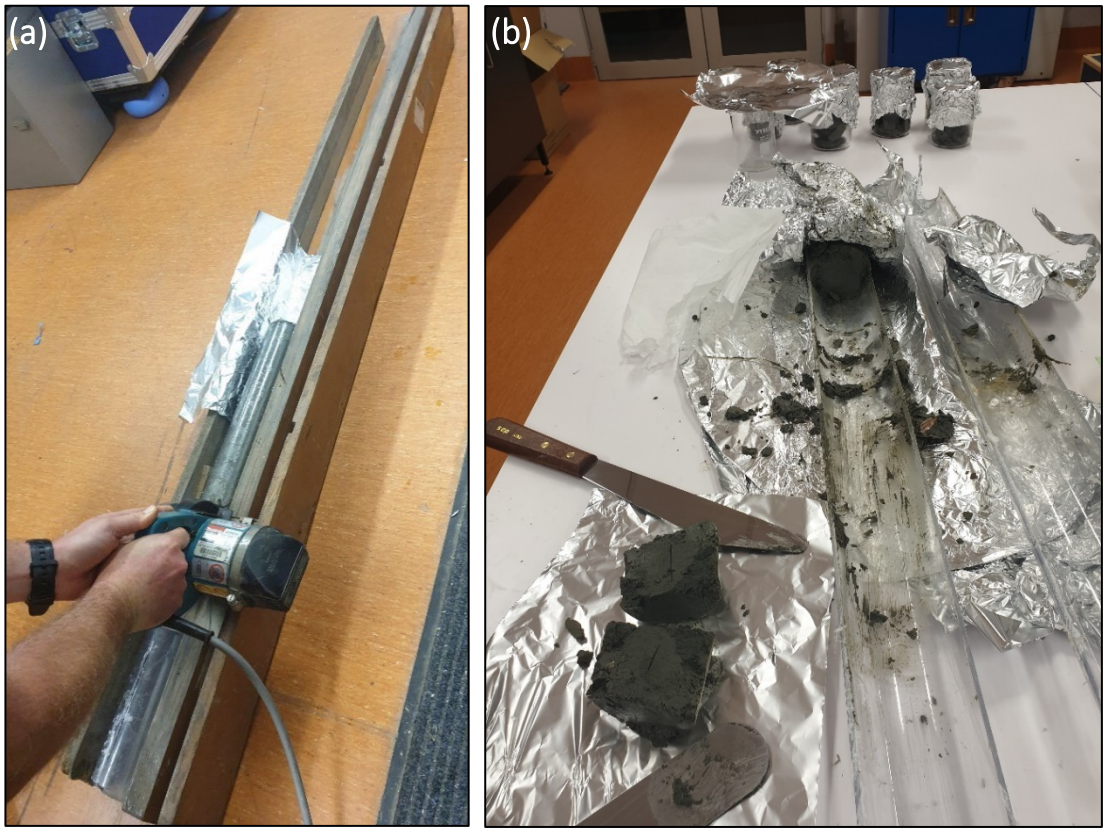

Figure 3. a) Opening of the CAB tube core and b) subsampling the sediment core, using metal slicers and storing each sub-sample into glass beakers.

Laboratory material decontamination

The following cross-contamination risk reduction measures were undertaken:

i. Intense rinsing of all laboratory material used with filtered water (1 µm filtered tap water)
ii. Glass and metal laboratory were preferentially used over plastic and consumables were used directly from sterile packaging.
iii. Equipment was kept covered with clean aluminium foil when not in use and the samples were covered as much as possible to minimize exposure risk.

iv. No synthetics - use a 100% cotton lab coat and avoid wearing synthetic clothes beneath the lab coat. Also, whenever possible, record the colour of the clothes worn underneath the lab coat as a precaution.

v. Daily controls – use glass microfiber filters (i.e. Cellulose filter) to monitor airborne particles (placing filter in a labelled open petri dish during laboratory work), and filtered tap water content (filtering 1L of water through the filter and conduct visual sorting in microscope).

vi. Decontamination of glass material – pre-clean all glassware before use with filtered tap water.

vii. Procedural blanks – procedure blanks were done in parallel to sample processing, following the same steps of the sample treatment, being the main difference the fact that they are run without the sample itself.

Pre-treatment of samples

Samples were pre-treated with a chemical or enzymatic digestion for destroying the organic matter (O.M.). Due to the strong reaction of plastics with high concentrated H_2O_2 solutions, a diluted solution is recommended *(Frias et al. 2018)*.

A solution of 10% H_2O_2 was used to digest the O.M., with an exposure time of approx. 48h (covered with clean aluminium foil) to make sure all the O.M. (Figure 4).

Figure 4. *Glass beakers with samples during digestion of O.M. Beakers were covered with aluminium foil to protect the samples from airborne microplastic contamination during digestion process.*

Density separation

The density separation process was used to separate the microplastic particles from the sediment grains. Most common plastics have very low densities, ranging from 0.8 to 1.4 g/cm^3; relative to densities for fine marine sediments (clay and silt), which are 1.4 – 1.55 g/cm^3.

This density difference is used to separate the lighter plastic particles from the heavier sediment grains by mixing the sediment sample with a saturated solution and stirring it for a prescribed amount of time *(Hidalgo-Ruz et al. 2012).*

The high-density solution (1.8 g/cm^3) sodium iodide (NaI) was used here for separating plastics from the sediment samples. Although it is one of the most expensive solutions (*Frias et al., 2018*), it has the advantage that it is the one with the highest density, increasing the efficiency of the density separation process for microplastic particles from the sediment grains. Whenever possible, the NaI solution was recycled and reused (loss of 35.9% after 10-times used *(Kedzierski et al. 2017).*

After O.M. digestion is finished, the sediment sample is mixed with the NaI solution, in a separating glass device (Figure 5), built at the University of Auckland following the design described in *(Nakajima et al. 2019*). The solution was stirred and mixed, using a metal stirring rod, for approximately 10 minutes to ensure full mixing of the sediment particles with the NaI solution (Figure 5).

The mixture was then left covered with clean aluminium foil to allow the sediment to settle to the bottom, while the low-density plastic particles remain in suspension or flow at to the surface of the NaI solution (Figure 5).

After separation is finished (for marine sediments this takes between 30 minutes and approximately 12 hours), the supernatant that includes the plastic particles was extracted to a new clean beaker for filtration.

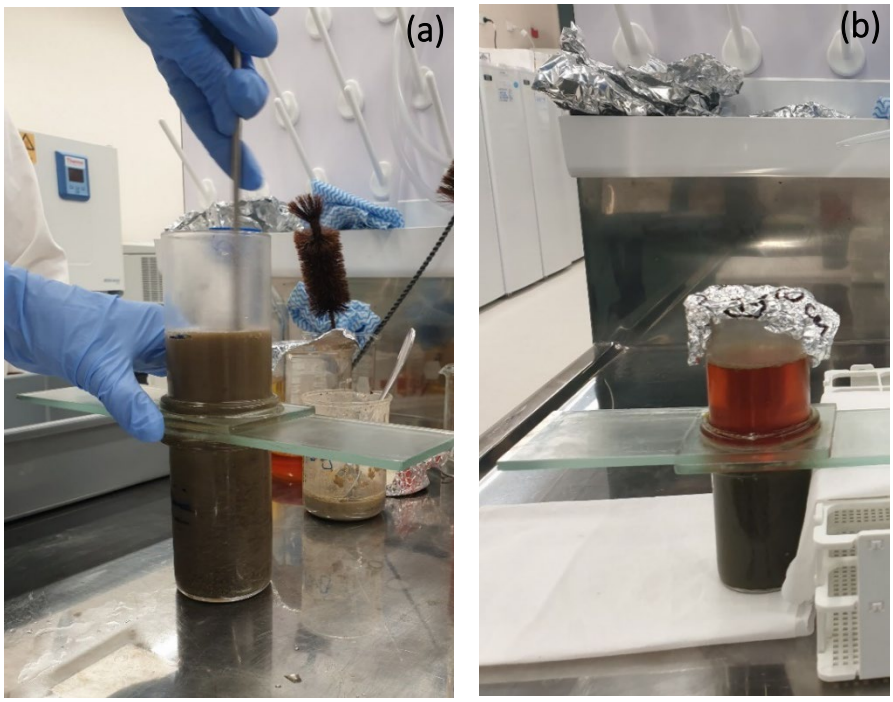

*Figure 5. a) Mixing sediment with NaI solution and **b)** separation process of sediment settling int the bottom while plastic particles and supernatant remain in suspension.*

Filtration

The solution of supernatant and plastic particles was extracted through filtration process (Figure 6).

A filtration kit was used with a fibre glass filter (Advantec Sterile Mixed Cellulose Ester Gridded Filter) with a pore size of 0.45 µm. After placing the solution into the filtration funnel, the beaker walls and the walls of the filtration device were washed and rinsed to ensure that all plastic particles are recovered on the filter. The filters were stored in a glass petri dish and kept drying in room temperature and in the dark (e.g., inside a drawer) to reduce light and airborne contamination.

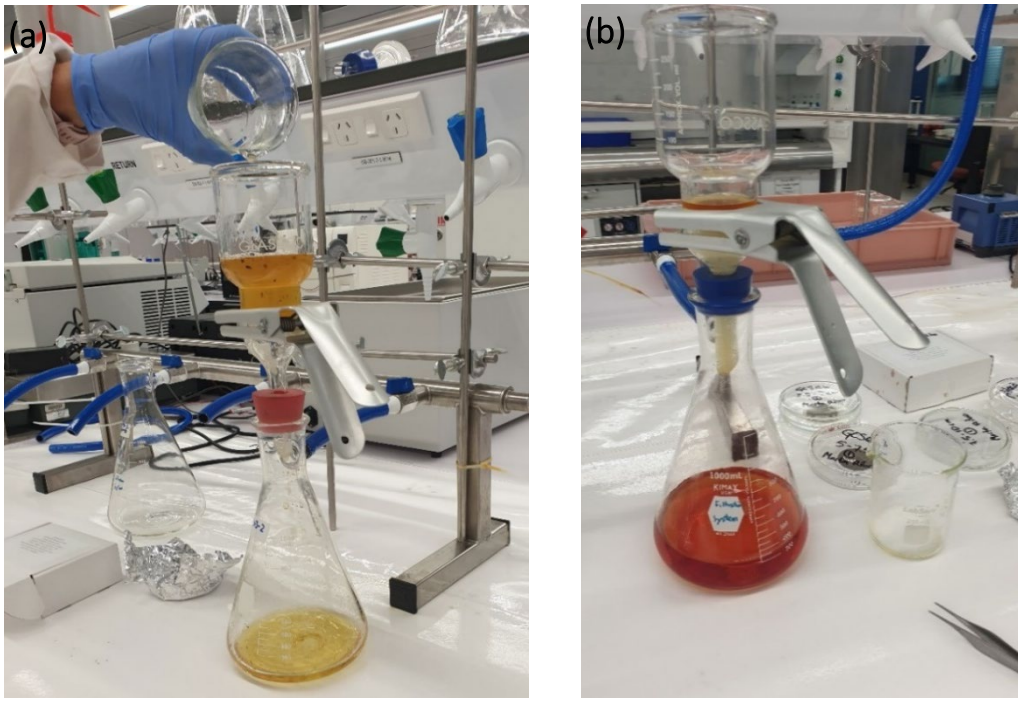

*Figure 6. Filtration process for plastic particles recovery (Note: the different colour of NaI solution between **(a)** and **(b)** is due to the reaction of NaI with atmospheric oxygen, this does not impact the effectiveness of the NaI solution when separating microplastics from sediment grains).*

2. Visual sorting

The obtained filters in the sample preparation were examined through a microscopic visual identification technique. Plastic particles were identified based on their physical characteristics using a Nikon SMZ 800N microscope, with 4x magnification (Figure 7).

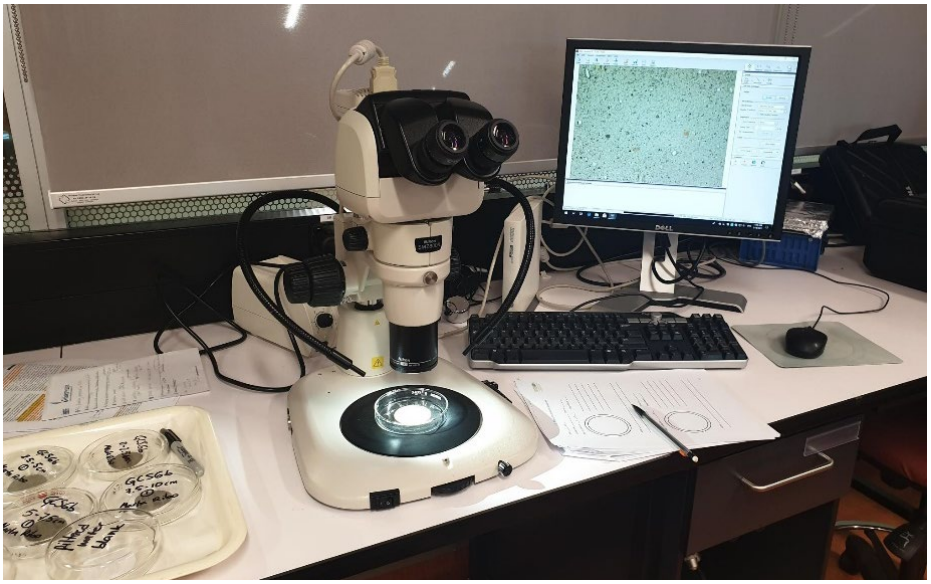

Figure 7. Setting of the Nikon SMZ 800N microscope at the University of Auckland Ecology Laboratory.

The criteria used during the identification of plastic particles involved the physical properties of the particles, including (*Frias et al. 2018*):

i. *Size*

Microplastics are defined as plastic particles < 5 mm, however particles ≤ 1 mm is the predominant class size identified in other the marine environments (*Kane and Clare 2019*). Two main size ranges are defined:

- Large microplastics: 1 to ≤5 mm
- Small microplastics: 1 µm to 1000 µm (=1 mm)

ii. *Type*

The most common microplastic types include: Fibre; Fragment; Pellet, Rope and filaments; Film.

iii. *Colour*

This criterion is relevant when aiming to identify factors of geographic influence and/or impact on local human activities. The most common colours are: Blue; Black; White or Transparent; and Red.

(note: the difference between white and transparent is opacity, white being opaque and transparent being translucent).

Results and discussion

The reporting microplastic identification and quantification results from this study was made using units of number of microplastics accumulated in each depth of the sediment cores (Table 1). When needed, for comparison with other studies, results can also be reported as number of microplastics per volume of sediment (# particles cm^3), considering the volume of each sub-sample (see details in sample separation in Methods section).

Table 1. Number of plastic particles in each depth subsection of the sediment core sample collected in the Anchorage and Marine Reserve sites (for location see Figure 1), for graphic representation of these data, see Figure 8.

Sediment core depth (cm)	Number of plastic particles
Anchorage	
0 – 2.5	8
2.5 - 5	2
5 – 7.5	5
7.5 – 10	15
10 – 15	23
15 – 20	16
20 – 25	9
25 – 30	8
30 – 35	26
35 – 40	8
Marine Reserve	
0 – 2.5	22
2.5 - 5	19
5 – 7.5	6
7.5 – 10	8
10 – 15	4
15 – 20	3
20 – 25	3
25 – 30	5
30 – 35	3
35 – 40	12
40 – 46	6

Microplastics were found in both sampling sites and present throughout the entire sediment core. The greatest abundance was in the top 5 cm in the Marine Reserve (Table 1 and Figure 8). However, at the Anchorage sampling site, the highest number of plastic particles were found at depths between 7.5 and 20 cm (Table 1 and Figure 8).

This results suggest there is a potential correlation between the plastic particle accumulation in the seafloor and the disturbance of the seabed due to human activities, as previous studies have shown that the Anchorage site is highly impacted by human activities, compared to the Marine Reserve, where there is low human footprint (*Watson et al. 2020*).

Repeat excavation by ships and anchors may explain why we observe higher plastic concentrations below the surface at the Anchorage site. Although microplastic particle concentrations at the Marine Reserve site mostly decrease with depth, higher concentrations at 35 to 46 cm could be attributed to sediment overturning by currents, bioturbation or previous scallop dredging activities.

Overall, variation was observed in abundance of microplastic particles across the sediment depth profile in both sediment cores collected (Figure 8). However, there is a clear difference between the two sampling sites, with the Anchorage site showing significant increases of plastic accumulation in the middle and lower parts of the sediment core (Figure 8a), while in the Marine Reserve location there is a predominant decrease of plastic particles with depth (Figure 8b).

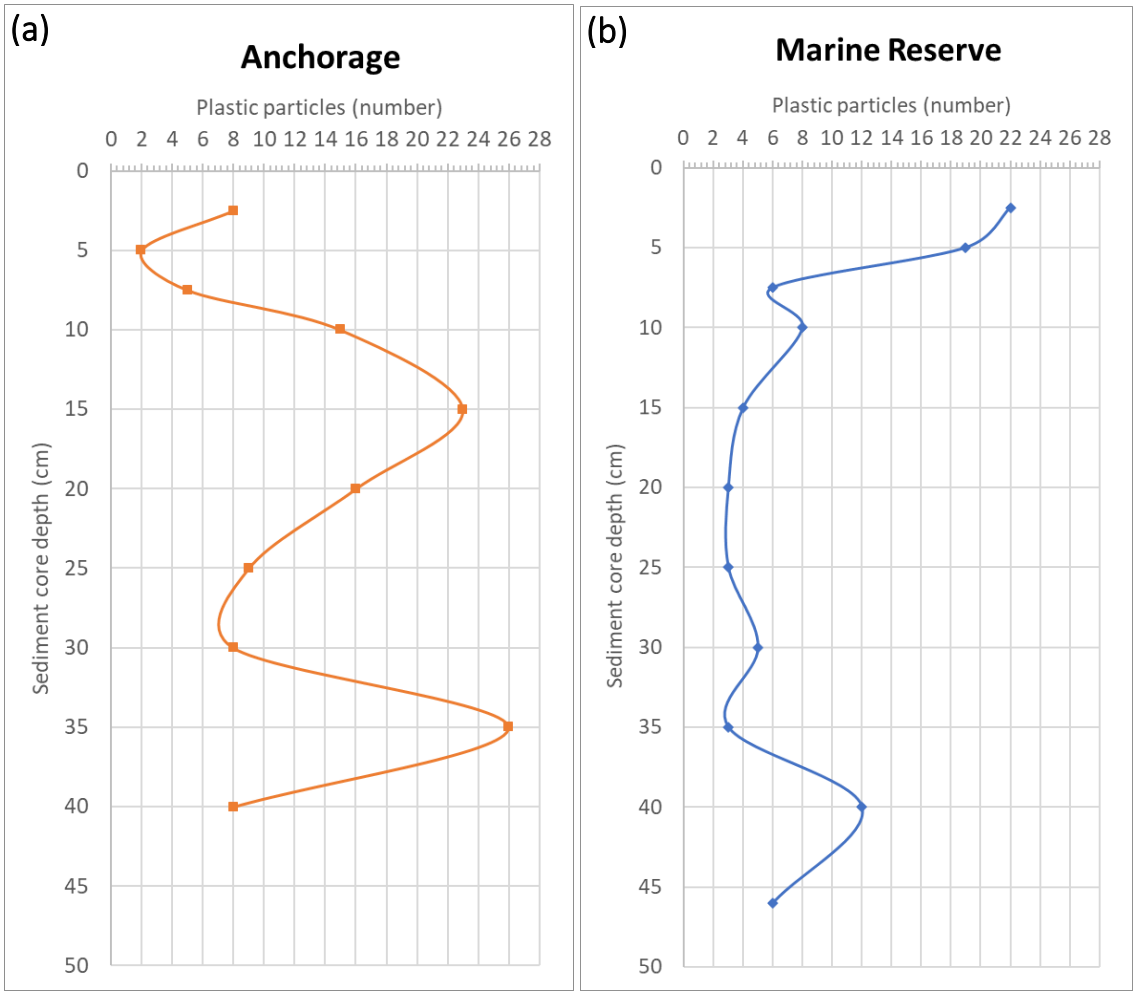

*Figure 8. Microplastic particles abundance at each depth interval averaged across the sediment cores collected in the **a)** Anchorage and the **b)** Marine Reserve (for location see Figure 1; for tabulated data points see Table 1).*

Overall, the observed plastic particles include both, large and small microplastics sizes. The most common types found in both sampled sites were fibres, fragments and pellets; and the predominant colours were blue, black, red, white and transparent (Figure 9).

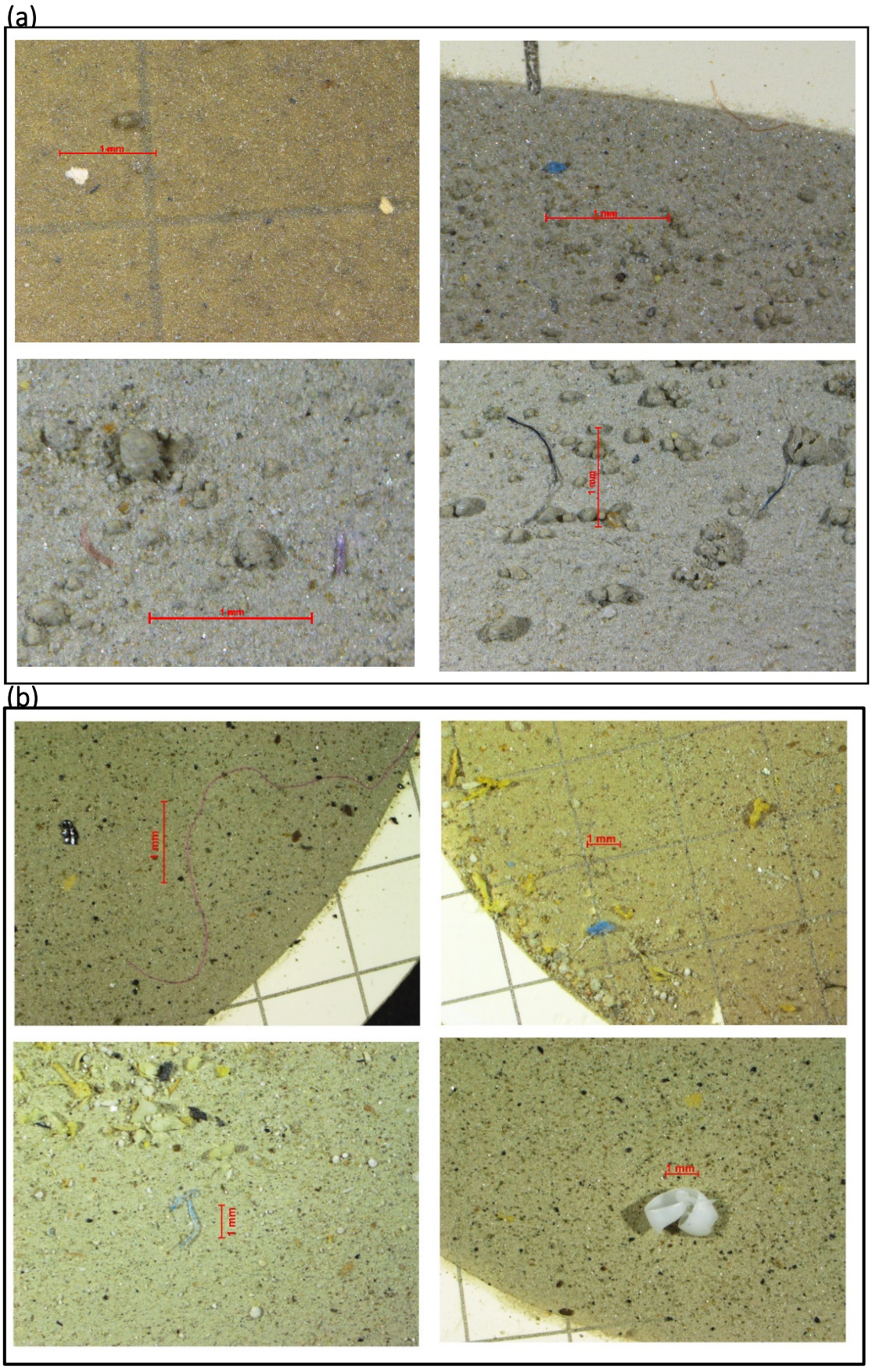

Figure 9. Images of microplastics identified in microscope when analysing the sediment samples of the ***a)*** *Anchorage and the* ***b)*** *Marine Reserve (see locations in Figure 1). Red lines indicate 1mm size.*

It is worth noting that at the Anchorage site (see location in Figure 1), higher variety of microplastic particles were found, including purple fragments (lower left panel, Figure 9a) and yellowish foam (upper right panel, Figure 9a), only observed in this site. The range of colours, types and sizes of microplastic particles suggest that there are numerous sources of plastic pollution accumulating in different areas within the QCS (Fig. 10; see Annex 1 for detailed information).

Differences in the distribution of plastic type and colour between the two sites suggest the geographical distribution of microplastic particles varies substantially within the QCS and likely reflects a combination of proximity to plastic source and sediment mechanisms transport (currents). One of the fibres identified in one of the subsamples, presented a degradation of colour, from dark blue to transparent (bottom right panel, Figure 9). This could be a result of degradation of the plastic particle caused by the H_2O_2 solutions during the O.M. digestion process (see details in pre-treatment of samples, in the Methods section).

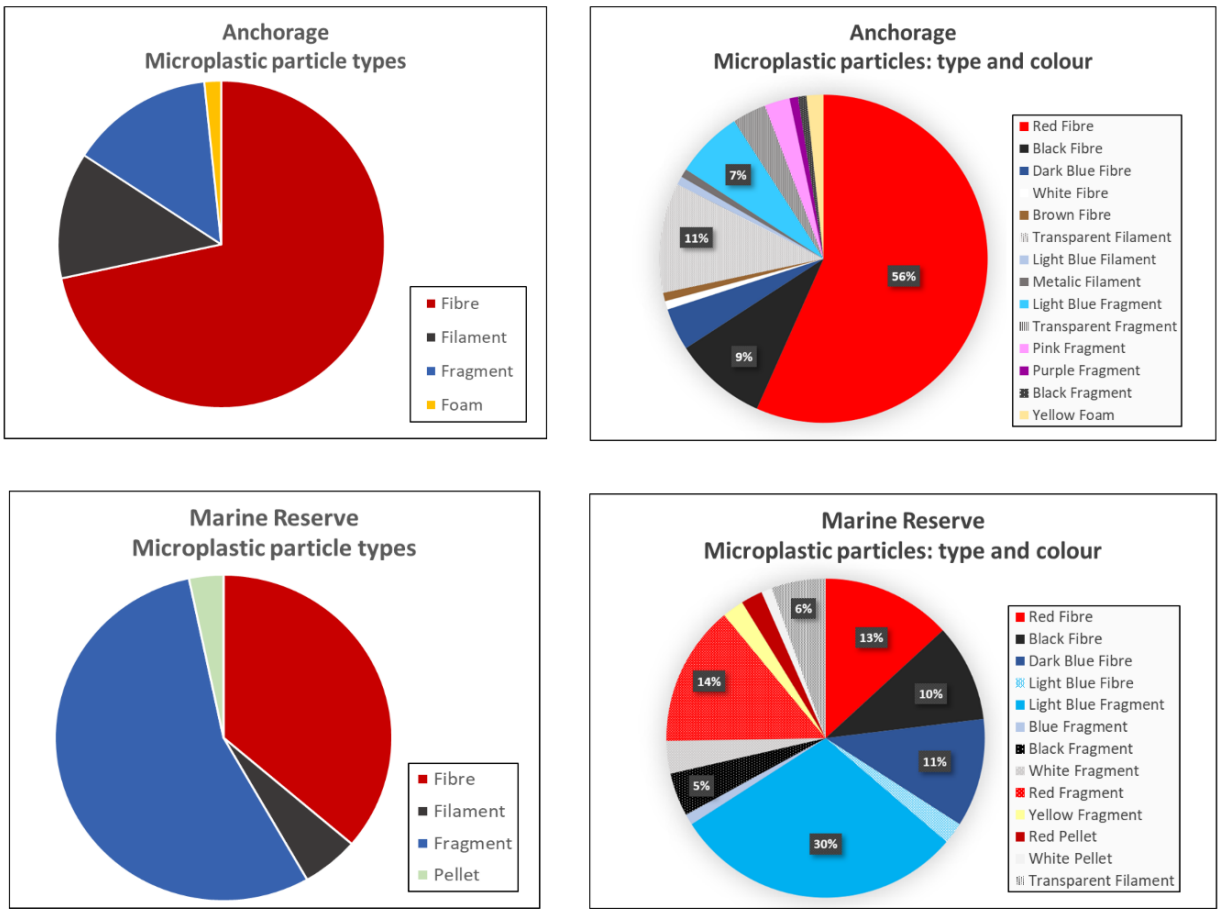

Figure 10. Microplastic particles classification considering type and colour for the Anchorage and Marie Reserve Sites (see locations in Figure 1).

Without plastic polymer identification analyses, conducted by Raman spectroscopy, Fourier Transformed Infrared (FTIR) spectroscopy or Pyrolysis-gas chromatography-mass spectrometry (Py-GCMS), as the most common methodologies, is extremely difficult to confidently determine with precision the source of the plastic particles. Thus, further research is needed to categorise the plastic particles through the detection of the chemical properties.

Finally, filters used during the daily controls (i.e., monitoring of airborne particles and content of filtered tap water) were examined. Unexpectedly, microplastics were found in the filters were tap water was filtered (Figure 11). These observations suggest some cross-contamination could potentially occurred during the laboratory analyses; however, it should be considered that the tap water filtered was from the University of Auckland laboratory, thus, cross-contamination is only considered for the microplastics of similar size, colour and type observed in found in both, the sediment samples and the filtered tap water filters.

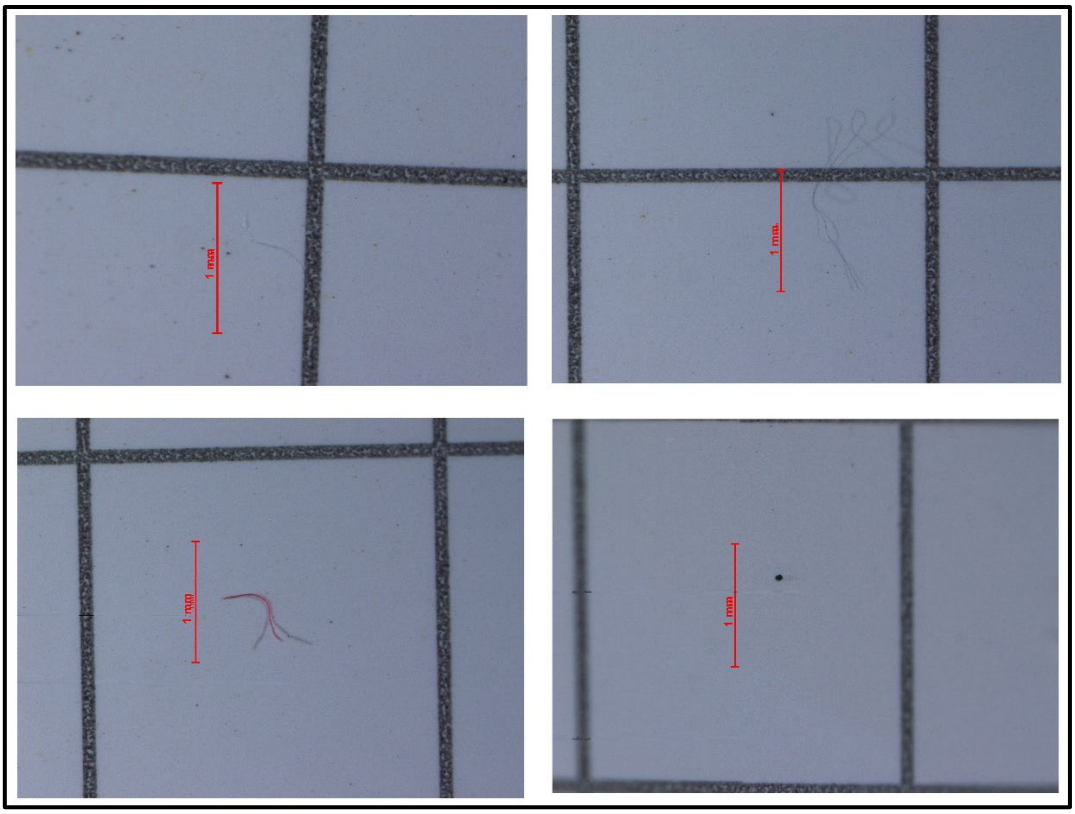

Figure 11. Images of microplastics identified in microscope when analysing the filters used for quality control on filtered tap water content. Red lines indicate 1mm size.

Conclusions

This study represents the first investigation into the presence and distribution of microplastic particles in marine sediments in Aotearoa/New Zealand. We observe microplastic particles within marine sediments in both the high-human impact site (Picton Anchorage) and in the near-pristine site (Marine Reserve), throughout both sediment cores, and in depths up to 50 cm below the seabed. The highest concentrations of microplastics were found at the Anchorage Site in depths between 10-15 cm (n=24 particles).

The Anchorage site is characterised by high concentrations throughout the entire length of the sediment core. Whereas, the Marine reserve site is characterised by high concentrations of microplastics in the uppermost surface layers and decreasing (for the most part) with depth below the seabed. We suggest high concentrations of microplastics in sediments below the seabed within the Anchorage site may be attributed to repeated overturning of sediments by anchor use, and more prolonged and proximal sources of plastic (i.e., near the largest coastal population within the QCS).

The Anchorage site also has a greater range of sizes, types and colours of microplastic, suggesting there are more sources of plastic to this region. Microplastics within the marine reserve site may be locally sourced (e.g., by recreational/commercial fishing) and/or transported to the marine reserve via ocean currents. Higher concentrations of plastics within the uppermost sediment layers could indicate increases in the use of microplastics in more recent years.

Surprising results were obtained when analysing the filters used as laboratory quality control, when filtering tap water, which was used to clean the laboratory equipment. Transparent and red microplastic fibres in addition to a metallic fibre and a black plastic fragment. These observations suggest there might have been some cross-contamination during the laboratory analyses. However, It should be taken into consideration that the tap water used was from the University of Auckland and not from the Marlborough Sounds area, therefore, contamination would be only considered when the same size, type and colour of microplastic are found in both, the sediment samples and the filtered tap water filters.

Future analyses should include the chemically characterising microplastic types (i.e., polymers) to correlate them with potential sources. Additional core samples targeting locations near potential plastic sources (e.g., marine farms, wastewater pipelines etc.) will also assist in identifying the dominant sources and transport of different microplastics within the marine QCS region.

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Annex 1

***Table 1**. Detail of Type, Colour and Size of the microplastic particles identified in the Anchorage site (for location see Figure 1)*

Anchorage				
Sediment core depth (cm)	Type	Colour	Size (L=large; S=small)	Amount
0 – 2.5	Fibre	Black	L + S	2
	Fibre	Dark Blue	S	2
	Fibre	Red	L + S	3
	Filament	Transparent	S	1
2.5 - 5	Fibre	Red	S	1
	Filament	Transparent	S	1
5 – 7.5	Fibre	Red	S	5
7.5 – 10	Fibre	Red	L + S	12
	Fibre	Black	S	1
	Fragment	Light Blue	S	2
10 – 15	Fibre	Red	L + S	16
	Fibre	Black	S	1
	Fibre	White	S	1
	Filament	Transparent	L + S	1
	Fragment	Transparent	S	4
15 – 20	Fibre	Red	L + S	8
	Fibre	Dark Blue to transparent	L	1
	Fibre	Black	L	2
	Filament	Transparent	L + S	3
	Fragment	Pink	S	1
	Fragment	Purple	S	1
20 – 25	Fibre	Red(ish) to Brown	S	5
	Fragment	Light Blue	S	3
	Filament	Light Blue	S	1
25 – 30	Fibre	Blue	L	1
	Fibre	Black	S	2
	Fragment	Light Blue	S	3
	Fragment	Black	S	1
	Filament	Transparent	S	1
30 – 35	Fibre	Red	L + S	17
	Fibre	Black	S	2
	Fibre	Dark Blue	S	1
	Fibre	Brown	S	1
	Filament	Transparent	S	3
	Foam	Yellow	S	2
35 – 40	Fibre	Red	S	1
	Fibre	Black	L	1
	Fragment	Pink	S	2
	Filament	Transparent	L + S	3
	Filament	Metallic	S	1

Table 2. Detail of Type, Colour and Size of the microplastic particles identified in the Marine Reserve site (for location see Figure 1)

Marine Reserve				
Sediment core depth (cm)	Type	Colour	Size (L=large; S=small)	Amount
0 – 2.5	Fibre	Red	L + S	9
	Fibre	Black	S	5
	Fibre	Dark Blue	S	2
	Fragment	Light Blue	L + S	3
	Filament	Transparent	L + S	3
2.5 - 5	Fibre	Dark Blue	L + S	2
	Fibre	Light Blue	S	2
	Fragment	Light Blue	S	14
	Filament	Transparent	S	1
5 – 7.5	Fibre	Red	S	2
	Fragment	Red(ish)	L+S	4
7.5 – 10	Fragment	Light Blue	L + S	4
	Fragment	Red	S	1
	Fibre	Dark Blue	S	2
	Filament	Transparent	S	1
10 – 15	Fragment	White	S	1
	Fragment	Yellow(ish)	S	2
	Fragment	Red(ish)	S	1
15 – 20	Fibre	Dark Blue	S	1
	Pellet	Red	S	2
20 – 25	Fragment	Blue	S	1
	Fragment	Red	S	1
	Pellet	White	S	1
25 – 30	Fragment	Black	L + S	3
	Fibre	Black	S	2
30 – 35	Fragment	Black	S	1
	Fibre	Black	S	2
35 – 40	Fragment	Light Blue	S	6
	Fragment	White	L + S	2
	Fibre	Dark Blue	S	3
	Fibre	Red	L	1
40 – 46	Fragment	Red(ish)	S	6