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RESEARCH ARTICLE

Investigation of microplastic pollution in Arctic fjord water: a case study of Rijpfjorden, Northern Svalbard

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Abstract

Microplastic contamination is an emerging issue in the marine environment including the Arctic. However, the occurrence of microplastics in the Arctic fjords remains less understood. Sample collections were conducted by trawling horizontally in surface water (0–0.4-m depth) and trawling vertically in the water column (0–200-m depth) to investigate the abundance, composition, and distribution of microplastics in the Rijpfjorden, Northern Svalbard, in the summer of 2017. Laser Direct Infrared chemical imaging technique was applied for the counting and identification of microplastic particles. A total of 1010 microplastic particles and 14 mesoplastics were identified from 41,038 particles in eight samples from the Rijpfjorden. The abundance of microplastics larger than 300 µm was 0.15 ± 0.19 n/m^3 in surface water, and 0.15 ± 0.03 n/m^3 in the water column of the Rijpfjorden. The microplastic particles identified in Rijpfjorden water consisted of 10 types of polymers. The dominant microplastics are polyurethane, polyethylene, polyvinyl acetate, polystyrene, polypropylene, and alkyd varnish. Historical ship activities and newly melted sea ice might be major sources of microplastics in the seawater of Rijpfjorden. In general, contamination of microplastics larger than 300 µm in Rijpfjorden water is at a low level in comparison to other polar waters. Further research is needed to confirm the origin and fate of microplastics below 300 µm in Arctic fjords.

Keywords Arctic water contamination · Zooplankton · Vertical profile · LDIR chemical imaging

Introduction

Microplastic pollution in the Arctic has been of great concern since the microplastic heritage was found to be released as the Arctic ice melts (Obbard et al. 2014). Sea ice is now considered a temporary and local source of microplastics in the Arctic (von Friesen et al. 2020; Peeken et al. 2018) in addition to other local sources, e.g., fisheries, shipping, and wastewater (e.g., Grøsvik et al. 2018; Herzke et al. 2021; Tekman et al. 2017), and to distant sources, e.g., lower latitudes via the atmosphere (Bergmann et al. 2019) and ocean currents (e.g., Cózar et al. 2017; Galgani et al. 2015). Since that discovery, microplastics have been found in many Arctic compartments, including sediment (e.g., Collard et al. 2021; Tekman et al. 2017), biota (e.g., Collard and Ask, 2021; Iannilli et al. 2019), and seawater (e.g., Huntington et al. 2020). In 2015, the first report on microplastics larger than 300 µm in surface and sub-surface seawater of Svalbard (Lusher et al. 2015) confirmed that Arctic seawater is contaminated with microplastics and also suggested to study the effects of microplastic-biota interaction in high

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zooplankton abundance areas. Furthermore, the abundance of microplastics in polar mixed layers in the Arctic Central Basin was higher than that in deep and bottom seawater and Atlantic water (Kanhai et al. 2018). A high abundance of anthropogenic microparticles (including microplastics) was detected in sea ice along the transect originated from Rijpfjorden (von Friesen et al. 2020) suggesting a local source of microplastics in that fjord. Such findings show the role of sea ice as a temporary microplastic sink besides source and pathways of transportation (Kanhai et al. 2020). However, little is known about the occurrence, composition, and sources of microplastics in the Arctic fjords.

The objectives of this study were first to characterize and estimate microplastic pollution in water of an Arctic fjord in Svalbard, and second to provide preliminary results on microplastic abundance and polymer types in different sampling types in Rijpfjorden waters.

Material and methods

Sample collection

We chose Rijpfjorden (~ 80°N, 22°E, ~ 40 km long, ~ 12 km wide), a typical Arctic fjord situated on the northern coast of Nordaustlandet in the Svalbard archipelago as a study area. Rijpfjorden is a shallow open fjord with one known deeper area (max depth 290 m) in the middle of the fjord (Fransner et al. 2017). Rijpfjorden is an S–N-orientated fjord extending onto the shallow shelf facing the Arctic Ocean and hence dominated by the cold water masses (Leu et al. 2011). Rijpfjorden is seasonally covered with ice (usually from October to July of the following year) (Soreide et al. 2010). Melting sea ice and runoff increased the amount of fresh water in the fjord. Water masses in Rijpfjorden mainly include transformed Atlantic water (TAW, with higher temperature and higher salinity), polar surface water (PSW, with higher temperature and lower salinity, influenced by melting and disintegration of glaciers and sea ice, etc.), intermediate water (IW, formed during the mixing process of PSW and TAW), and winter cooled water (WCW, high salinity, and low temperature) based on the field hydrographic data (Fig. S1) and by referring to Hop et al. (2019). However, there are indications that Rijpfjorden is turning into a warmer system with an increased inflow of Atlantic water and hence larger input of Atlantic zooplankton (Hop et al. 2019).

Microplastic collection in the Rijpfjorden was conducted on the Norwegian research vessel Lance during the Kongsfjorden-Rijpfjorden Cruise in August 2017 (Fig. 1 and Table 1). Our sample collection method followed that of Kovač Viršek et al. (2016). A Manta net (Hydro-bios, mouth opening 40 cm × 70 cm, mesh size 300 µm) was trawled

Fig. 1 Location of sampling sites (red open circles) along the Rijpfjorden transect and sample collection. Sampling net bags need to be cleaned by re-flushing with high-pressure water (a) before and after sampling by vertical trawl at sites R3 using WP2 net (b) and horizontal trawl at R1, R2 and R4 using Manta net (c). (Maps modified from TopoSvalbard, https://toposvalbard.npolar.no)

Table 1 Description of sampling sites, layers, and volumes

Site no	Longitude	Latitude	Water depth (m)	Sampling layer	Volume per sample (m^3)	Notes
R1	22°09.58′	80°08.42′	198	Surface water (0–0.4 m)	103.7	Visible plastics
R2	22°09.58′	80°10.18′	182	Surface water (0–0.4 m)	103.7	Visible plastics
R3	22°18.26′	80°17.10′	225	Water column (0–200 m)	51.03	Zooplankton-rich samples
R4	22°07.17′	80°39.14′	128	Surface water (0–0.4 m)	172.9	Algae-rich samples

horizontally with the speed of 1.2 knots at R1 and R2 and 2 knots at R4 for 10 min in the surface water (depth 0–40 cm). In addition, a vertical trawling net (WP2, HydroBios; opening diameter 57 cm /area 0.255 m^2, mesh size 200 µm) was applied for a particle sample collection from the water column (0–200-m depth). Duplicate samples were conducted at all sampling sites. The volumes of the water passing through the net were calculated by multiplying the towing distance by the opening area of trawling net. After trawling, the particles, phytoplankton, and zooplankton retained on the nets were backwashed by high-pressure seawater, and then the concentrates were filtered through a nylon filter membrane (60-µm mesh size, 47-mm diameter) on board, and then stored at 4 °C in the filter membrane cases.

Sample preparation

Our samples contained microplastic particles, phytoplankton, and zooplankton. Therefore, we adopted a preparation method from NOAA Marine Debris Program (Masura et al. 2015). Firstly, Fenton's reagent method (20 mL of 30% H_2O_2 and 20 mL of 0.05 mol/L Fe(II) solution) was used for organic material digestion, and the digestion process was repeated until visible natural organic substances in the liquid disappear completely. Then, NaCl powder (300 g/L) was added to the digested solution for the density separation step (solution density = 1.15 g/mL). The NaCl powder dissolved completely at 75 °C. After the wet peroxide oxidation and the density separation, the liquid containing suspended particles was filtered through a nylon filter membrane (60-µm mesh size). The density separator was rinsed with milli-Q water (18.2 MΩ·cm, 25 °C) to ensure all suspended particles were successfully transferred to the filters. Afterward, all the solid particles on the filters were rinsed with 10–15 mL of anhydrous alcohol for 5 times into glass vials (25 mL volume), and then the sample vials covered with aluminum foil cover were properly concentrated to about 1 mL in an oven at 55 °C and transferred to LC sample vial (Agilent, USA) by a glass dropper.

Particles larger than 0.5 mm were picked out with tweezer (Vetus, Shanghai, China) and stored in petri dishes for further analysis. Then the particles were observed under a continuous phototype microscope (SM362, Shangguang New Optical Technology Co., Shanghai, China), photographed, and measured with Toup View software (Ver.3.7, Atlas Optoelectronics Co., LTD., Hangzhou, China).

Microplastic enumeration and identification

Size and polymer types of large microplastics (i.e., > 500 µm) were identified individually by the Fourier transform infrared (FTIR) technique with attenuated total reflectance module using Agilent Cary 630 FTIR. Laser Direct Infrared (LDIR) chemical imaging system (Agilent 8700) was used to enumerate and identify microplastics in equivalent sphere diameters below 500 µm (the smallest particle diameter detectable with the LDIR method is 20 µm), by following the principles and test procedures described by Scircle et al. (2020) and Li et al. (2021). Particle scans and the Agilent Clarity Software conducted all polymer identifications in real time. A matching threshold was set at 80%. Except for unknown particles with a very low matching degree, the identified non-plastic substances included mostly natural cellulose, coal, silicate, and other natural substances.

The particle size of all microplastics in this study is calculated as the equivalent spherical diameter based on the following equation:

$$D = 2 \times \sqrt{\frac{S}{\pi}} \quad (1)$$

where S is the particle area scanned by the LDIR imaging system; D is equivalent spherical diameter, the particle size used in this study.

In this study, we provided detailed results on microplastics larger than the mesh size of sampling nets (300 µm for the mesh sizes of Manta net and 200 µm for the WP2 net).

Quality control

A variety of measures for avoiding contamination and assessing quality control were adopted in this study. This study did not have field blanks but three measures were done to limit field contamination as far as feasibly possible. Firstly, backwashing nets before and after each sampling action are very important. We flushed the trawling nets with a high-pressure water gun to ensure that the residual microplastic particles on the net meshes were fully removed, and there was no cross-contamination between samples. Secondly, the trawling nets were extended through the L-shaped boom to the water about 5 m away from the ship to collect microplastics. And we tried to trawl in line with the direction of the ship. Thirdly, the microplastics backwashed from the sampling net samples were quickly recovered by 60-µm filters, so it could reduce the time for sample processing on the ship, and then reduce the contamination onboard. As for processes in the laboratory, glass containers and Milli-Q water were used to prepare the solutions, and once the solutions met open-air, covers of aluminum foil were used to prevent airborne contamination. The containers used in the experiment were rinsed with Milli-Q water 3 to 5 times. All the solvents were filtered by Whatman GF/F filter paper before being used in the oxidation and density separation process. In addition, all the laboratory operations were carried out wearing cotton lab coats to prevent contamination. A reagent

blank was set up using pre-filtered anhydrous alcohol during the sample preparation for identification.

Results

Abundance

Only one PET particle (> 300 µm) was detected in the reagent blank and was deducted from the subsequent sample data. Fourteen mesoplastic particles (5–20 mm) were found in the shapes of fibers, sheets, and fragments (Fig. S3 and Table S1). A total of 1,010 microplastics (< 5 mm) including 49 fibers (Length/Diameter > 3) were identified by using LDIR among 41,038 particles in eight samples from the Rijpfjorden seawater (Table 2). One hundred thirty-three microplastics (larger than the corresponding sampler mesh size) with 14 fibers (length/diameter > 3) were detected by LDIR, while 107 of the microplastics were larger than 300 µm, the largest mesh size of the sampling nets used in this study. The abundance of microplastics larger than 300 µm was 0.15 ± 0.19 n/m^3 in surface seawater with a maximum observed at the bay head (0.55 n/m^3 of R1-1), and 0.15 ± 0.03 n/m^3 in the water column at R3 (0.41 ± 0.17 n/m^3 of microplastics larger than 200 µm). The lowest abundance of microplastics in Rijpfjorden seawater was observed at the fjord mouth (R4), with no microplastic particle larger than 300 µm. There was shown no significant difference between the number of microplastics found in surface water and water column.

Particle size distribution

Microplastic particle sizes ranged from 200.1 to 4694.1 µm in the different samples (Fig. 2). About 79.7% of the microplastics identified were in the size of 300–5000 µm while 20.3% in 200–300 µm. R1 had the widest particle size distribution range of microplastics (321.1–4694.1 µm), followed

Table 2 Quantification of particle analyzed and microplastics identified from the Rijpfjorden seawater samples in different size ranges. Mean values ± standard deviation were calculated for the abundance of microplastics in surface water (a) and water column (b), respectively

Sample no	Particles analyzed (n)		No. of microplastics (n)		Microplastic abundance (n/m^3)	
	60–300 µm	300–5000 µm	60–300 µm	300–5000 µm	60–300 µm	300–5000 µm
R1-1	1807	2853	411	57	3.96	0.55
R1-2	10,865	88	63	21	0.61	0.20
R2-1	4022	133	41	11	0.40	0.11
R2-2	466	6	45	3	0.43	0.029
R3-1	5557	54	46	6	0.90	0.12
R3-2	6254	2768	171	9	3.35	0.18
R4-1	4184	13	83	0	0.48	0
R4-2	1963	5	43	0	0.25	0
Sum	35,118	5920	903	107	1.02 ± 1.44^a; 2.13 ± 1.73^b	0.15 ± 0.19^a; 0.15 ± 0.03^b

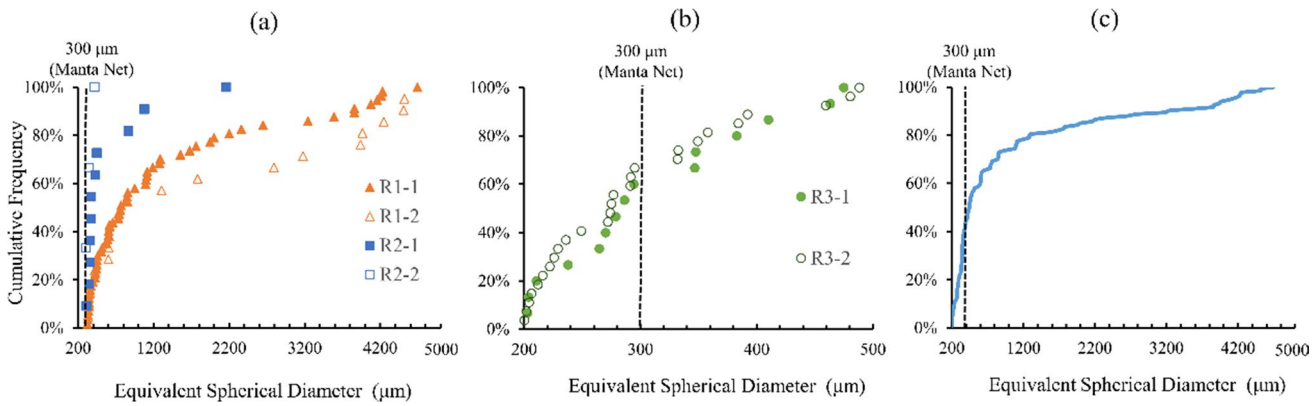

Fig. 2 Cumulative frequency distribution of microplastics at different sites/samples collected from the Rijpfjorden. (a) Microplastics detected in surface water samples, (b) microplastics larger than the mesh size of WP2 detected in water column samples, (c) all microplastics larger than the mesh size of the sampler (Manta Net or WP2) detected in this study. Lines are marked to distinguish the distribution of microplastics larger than the sampling net mesh sizes (300 µm for Manta Net). Particle size refers to the equivalent spherical diameter

by R2 (306.2–2159.9 µm), and R3 (200.1–488.3 µm) was the narrowest. The particle size distributions of microplastics in surface water and water column showed obvious differences. The rapid particle accumulation ranged from 200 to 1200 µm and 200 to 300 µm, and the microplastics distribution in water column turned to be more uniform than the microplastics in surface water (Fig. 2(a, b)). The particle size distribution of the microplastics in this study showed a near-linear accumulation between 200 and 600 µm, and the degree of accumulation became stable thereafter (Fig. 2(c)). Microplastics < 500 µm dominated the particle size in R2 and R3 samples with a ratio of 73 ~ 100%. In general, nearly 80% of the microplastics had a particle size between 200 and 1300 µm.

Polymer composition

The composition of microplastic particles was identified in a variety of polymers (Fig. 3): polyethylene (PE), polyurethane (PU), polyvinyl acetate (PVA), polystyrene (PS), polypropylene (PP), polyvinyl chloride (PVC), polymethyl methacrylate (PMMA), rubber, polyvinyl alcohol, and alkyd varnish (AV) were detected.

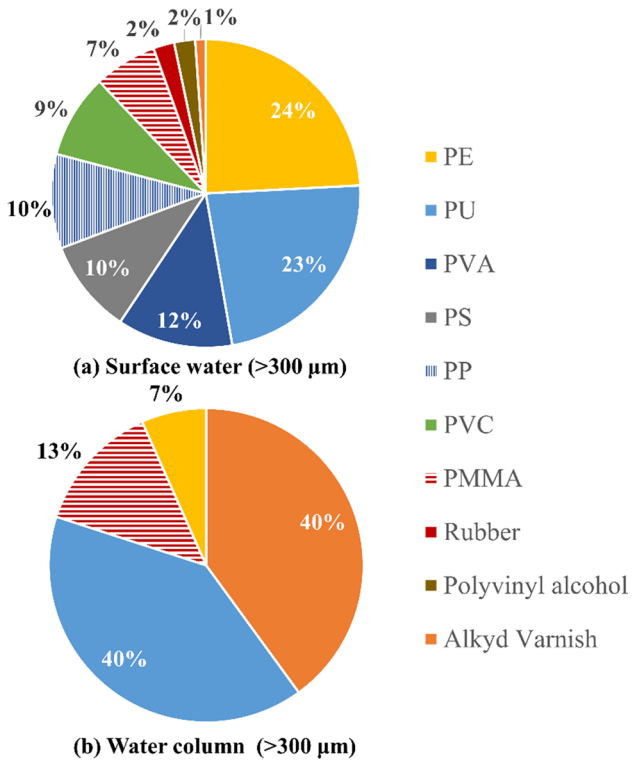

Fig. 3 Polymer composition of microplastics collected from the Rijpfjorden. (a) microplastics from surface water (> 300 µm), (b) microplastics larger than 300 µm from the water column. Ten types of polymer, i.e., polyethylene (PE), polyurethane (PU), polyvinyl acetate (PVA), polystyrene (PS), polypropylene (PP), polyvinyl chloride (PVC), polymethyl methacrylate (PMMA), rubber, polyvinyl alcohol, and alkyd varnish (AV) were detected

Typical LDIR chemical imaging spectra are shown in Fig. 4. Particle identification and spectrum matching were performed automatically during the operation of the LDIR system. All microplastic particles in this study had more than 80% matching degree with the microplastic spectrum in the spectral library.

The distribution of the polymer composition varied with the size range and the sampling methods. Specifically, PU, PE, and PVA were the 3 most abundant polymers accounting for 57% of all microplastics larger than 300 µm. The polymer composition of microplastics in surface seawater showed obvious differences from the water column (Fig. 3(a, b)). PU was dominant in the water samples from the two different compartments (surface water and water column). However, PE (24%) dominated in the horizontal trawl samples (Fig. 3(a)) while alkyd varnish (40%) dominated in the vertical trawl samples (Fig. 3(b)).

Discussion

Occurrence characteristics of microplastics in Rijpfjorden water

The contamination of large microplastics in Rijpfjorden seawater is comparable to those reported in several other Arctic studies using trawl-sampling methods (Tables 2 and 3). Firstly, the abundance of microplastics larger than 300 µm in surface seawater from Rijpfjorden (0–0.55 n/m^3) is similar to that from the fjord Nuum Kangerlua, Greenland (Rist et al. 2020), where the prevalence of microplastics in surface seawater sampled by bongo nets (300 µm mesh size) was on average 0.12 n/m^3 and also lies in the range of microplastics (0–1.31 n/m^3) in surface seawater of Southwest of the Svalbard archipelago (Lusher et al. 2015). Secondly, the levels of microplastics larger than 200 µm (0.29–0.53 n/m^3) or 300 µm (0.12–0.18 n/m^3) in the water column in our study are also among the range of those in sub-surface seawater of Southwest Svalbard (0–11.5 n/m^3, Lusher et al. 2015) and seawaters underlying the Arctic central basin ice floes (0–18 n/m^3, Kanhai et al. 2020). However, microplastic abundance in Rijpfjorden seawater was 1–2 orders of magnitude lower than that in other Arctic waters collected by small size mesh filters (< 10 µm), e.g., the Polar Mixed Layer in the Atlantic Ocean (13–501 n/m^3, Enders et al. 2015) and surface water in the fjord Nuup Kangerlua (67–278 n/m^3, Rist et al. 2020). Therefore, the contamination level of microplastics was generally underestimated by trawling nets for sample collection as most small size microplastic particles could pass through the net mesh (Lindeque et al. 2020; Tokai et al.2021). However, regardless of their sampling methods, the abundances

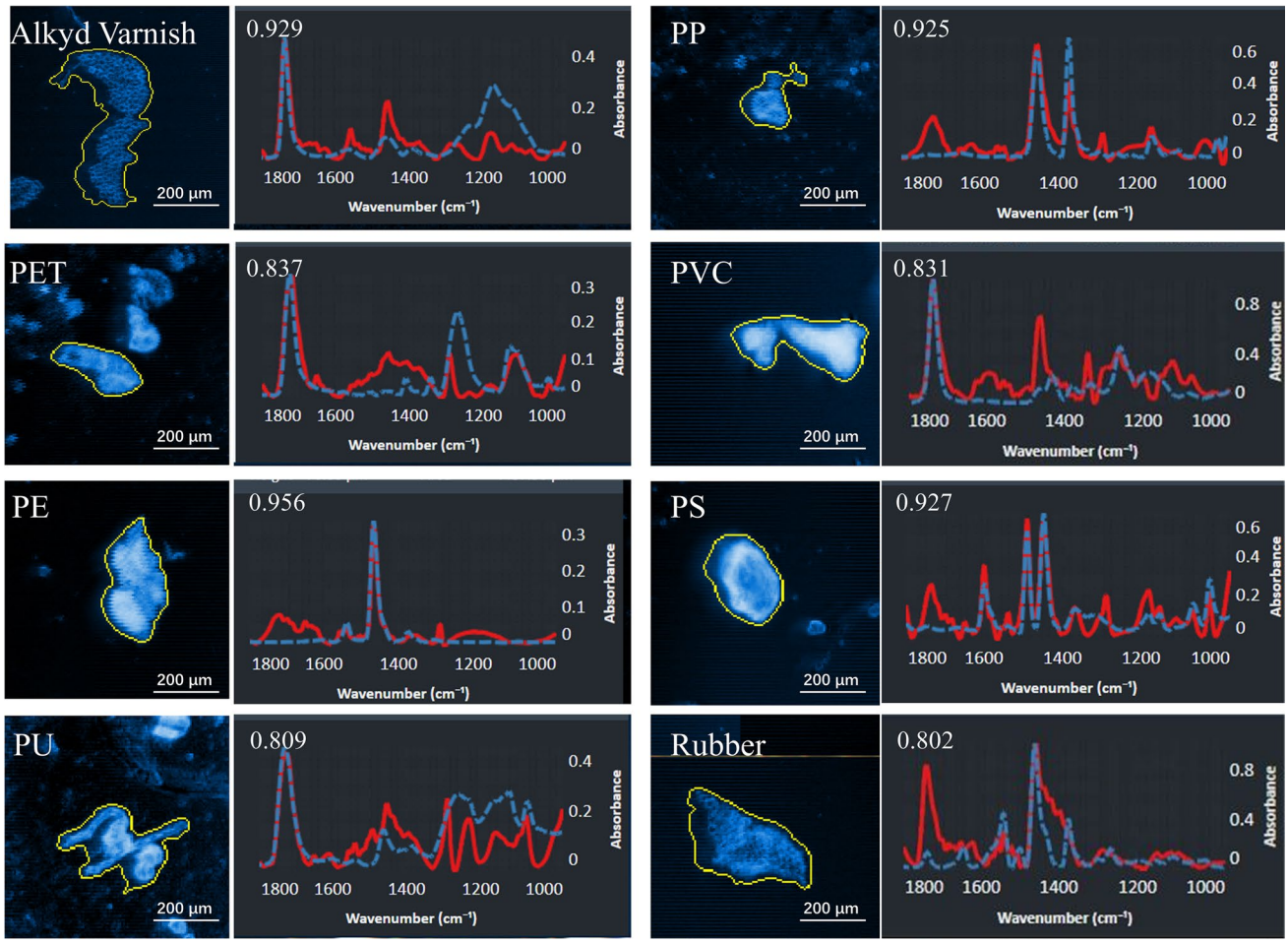

Fig. 4 Typical LDIR (laser direct infrared) chemical imaging spectra of eight microplastics identified as alkyd varnish, polyethylene terephthalate (PET), polyethylene (PE), polyurethane (PU), polypropylene (PP), polyvinyl chloride (PVC), polystyrene (PS), and rubber. Blue lines are reference spectra and red lines are measured ones. The number on the upper left of the spectrum is the degree of matching with the standard spectrum

of large microplastics in Arctic waters (Enders et al. 2015; Lusher et al. 2015; Rist et al. 2020) are relatively comparable. When we put all the 8 samples data together, we can conclude that the microplastic pollution in the Rijpfjorden seawater (0–0.55 n/m^3) was at a low level in Arctic waters reported (range from 0 to 26×10^7 n/ m^3 in Table 3).

In this study, we have found microplastics smaller than the mesh sizes of samplers, and this part accounted for a large proportion, which is consistent with the result found in the South China Sea (microplastics size < 300 µm contributed 92% of the total number of microplastics) (Cai et al. 2018). This is related to the use of the LDIR method (lower size limit of 20 µm) to detect microplastics, which can obtain more accurate small-size microplastic data than traditional methods.

The particle size distribution of microplastics in the water column had a narrower range than that in the surface water (Fig. 2). This phenomenon is consistent with the investigation conducted by Li et al. (2020), who suggested that it could result from the combined effect of hydraulic conditions, biofouling, and the properties of the MPs. Chubarenko et al. (2016) proposed that the surface area of fibrous microplastics allows biofouling easier than in spherical microplastics and the particles rapidly sink, which can explain 71% of microplastic fibers were found in the water column (Table S3).

The dominant polymers (PE, PU, PVA, PS, PP, PVC, Alkyd Varnish, PMMA) in this study were consistent with similar studies that investigated the microplastics in Rijpfjorden sea ice (Friesen et al. 2020) and surface water in Fjord Nuup Kangerlua (Rist et al. 2020). Polymer types are more abundant in surface water (10 types) than in water column (4 types) (Fig. 3), which possibly indicate the major sources from upper sea layer than deep water. Low-density microplastic PE was found in both water column samples. Since the water column also included the surface water in

Table 3 Global comparisons of the abundance and polymer composition of microplastics in the Arctic environment

Study areas	Methods (match degree)	Mesh size of nets[a] or filters[b] (μm)	Size (μm)	Abundance (n/m^3)	References
Sea ice					
Rijpfjorden, Svalbard	μFTIR (> 60%)	10[b]	50–5000	$(1.58 \pm 1.55) \times 10^{5\#}$	von Friesen et al. (2020)
Arctic central basin	μFTIR (> 60%)	250[a]; 1.2[b]	100–4990	$(2–17) \times 10^3$	Kanhai et al. (2020)
Fram Strait; the Central Arctic	Imaging FTIR	0.2[b]	11–500	$(0.3–26) \times 10^7$	Peeken et al. (2018)
Freshwater lake					
Ny-Ålesund, Svalbard	Raman microscopy; μFTIR; synchrotron radiation μFTIR	500[a]; 1.5[b]	NA	1680 ± 440 AMPs/m^2	González-Pleiter et al. (2020)
Sub-surface water					
Southwest waters of Svalbard	μFTIR	333[a]	250–7710	0–11.5	Lusher (2015)
Surface water					
Arctic central basin	μFTIR	250[a]; 1.2[b]	100–4990	0–18	Kanhai et al. (2020)
Fjord Nuup Kangerlua, Greenland	FPA-mFTIR-Imaging	10[b]	11–5000	67–278 (4.7–19.7)*	Rist et al. (2020)
Fjord Nuup Kangerlua, Greenland	FPA-mFTIR-Imaging	300[a]	11–5000	0.08–0.4	Rist et al. (2020)
Southwest waters of Svalbard	μFTIR	333[a]	250–7710	0–1.31	Lusher (2015)
Atlantic Ocean	Raman micro-spectrometry	300[a]; 10[b]	10–1000	13–501 (0.01–0.5)*	Enders et al. (2015)
Rijpfjorden, Svalbard	LDIR Chemical Imaging	300[a]; 60[b]	60–5000	0–0.20*	This study
Vertical water column					
Rijpfjorden, Svalbard	LDIR Chemical Imaging	200[a]	60–5000	0.12–0.18*	This study

AMPs anthropogenic microparticles, including microplastics

[#] AMPs/m^3

[*] Abundance of microplastics > 300 μm

the vertical trawl area during sampling, PE particles floating on the water surface at the top of the vertical water column could be captured. Lower-density microplastics (e.g., PP) were found in other investigations (Li et al. 2020; Tekman et al. 2020), indicating that density has a poor influence on microplastic displacement in the vertical water column, especially when hydrodynamics is complex and multifactorial there. It is even possible to find lower-density microplastics in Arctic sediment (Collard et al. 2021). Besides, fouling can increase the density of microplastics and contribute to the sinking process (Katija et al. 2017).

Challenges in the sampling of microplastics in Arctic fjord water

The quantification of microplastics in seawater was largely determined by the investigation methods, e.g., sampler/filter mesh size and identification technique (Table 3). About the prevalence and importance of microplastics in Rijpfjorden, the sample collection required a filter with a small pore size and thus made the filtration process more challenging. Moreover, due to the low abundance of large microplastics in Arctic waters, a much larger volume of water is needed compared with other regions.

In addition, both samples from R3 were dominated by zooplankton (including *Calanus glacialis* and *Clione limacina*) while both samples of R4 contained phytoplankton (Fig. S2). Consequently, the separation of microplastics from living organisms (e.g., phytoplankton, zooplankton) in samples may also be very challenging in productive Arctic waters. Another study performed in the Arctic also found out that subsurface waters were more contaminated than surface waters (Lusher et al. 2015). At present, there is no scientific publication of microplastic ingestion by planktonic organisms in Arctic waters, but it has been observed in Arctic benthic amphipods (Iannilli et al. 2019) whereas in a sand hopper from sandy beaches (Iannilli et al. 2018), and planktonic species in experimental studies (e.g., Cole et al. 2013; Vroom et al. 2017). Jones-Williams et al. (2020) indicated that microplastics, even at low abundance in Atlantic seawater, still have the potential to be encountered and consumed by amphipods. Given these facts, we suggest that

large zooplankton in Rijpfjorden could be exposed to microplastics in seawater.

A higher abundance of microplastics was noticed in the first sample (R1-1, Table 2) of this study. That difference might be partially attributed to the contamination of the net rinsed incompletely before sampling. This proves that a proper rinsing of sampling equipment is necessary between sampling sites and between replicates.

Potential inputs of microplastics in Rijpfjorden water

Sea ice melting may substantially release microplastics into seawater in Rijpfjorden compared with the study on microplastics in sea ice of Rijpfjorden at the same time (von Friesen et al. 2020) (Table 3). There was similar polymer composition of microplastics (e.g., PE and varnish/paint) between the sea ice and seawater of Rijpfjorden. Moreover, PU dominated in all water samples in this study, which is consistent with the studies on surface water and ice floes samples from the Arctic central basin (Kanhai et al. 2020). Peeken et al. (2018) observed a unique pattern of microplastic in sea ice from the Svalbard area and concluded that the Arctic sea ice is an important temporary sink and transportation media for microplastics. Since sea ice was observed at all sampling sites, microplastics in Rijpfjorden could be mainly originated from summer sea ice melting (von Friesen et al. 2020). Rijpfjorden normally opens up between mid-July and mid-August (Ambrose et al. 2006), which is the time our sampling was conducted. Although the ice floes will leave the fjord and drift towards the Arctic Ocean after they burst, they are often packed back into the fjord under the influence of the north wind (Leu et al. 2011) and melt inside the fjord which could release microplastics into seawater.

In addition, atmospheric deposition is another potential source because small particles are available for long-distance or even global transmission (Betzer et al. 1988). Bergmann et al. (2019) observed microplastics in snow from the Fram Strait (8.8 ± 7.9 n/m^2) and three different locations of Svalbard (1.4 ± 0.4 n/m^2) thus further strengthening the significant contamination of microplastics through air circulation and scavenged by wet deposition, long-distance transportation to the Arctic from urban regions, and scavenged by wet deposition on land and water surface.

Furthermore, the large proportion of PU (23% in surface water and 39% in water column) and varnish (49% in the water column) in water samples of this study could also imply that vessels could act as a source. PU is a commonly used thermal insulation material while the varnish is often used for ship protection. This source needs to be confirmed because it may closely be related to human activities such as cruises and fisheries (Stocker et al. 2020) in the Arctic region. If so, adapted measures should be taken as far as possible in the future to reduce that kind of input.

This study provides a basis for the sources of microplastics in the Arctic fjords with a limited amount of data. The identification of these sources and the content of microplastics in zooplankton as well as the impact on these polar organisms require further research.

Conclusion

Microplastic contamination occurred in the seawater of Rijpfjorden with various polymer types (such as PU, PE, alkyd varnish), but the abundance of microplastics larger than 300 μm was relatively low. No recent anthropogenic input of microplastics was observed in the Rijpfjorden, whereas there was some evidence that historical ship activities and newly melted sea ice might play a major role in the presence of microplastics in the seawater of Rijpfjorden. Key species of zooplankton might be exposed to microplastics in the zooplankton-rich water column. In addition, the LDIR chemical imaging technology used in this study has been proved to be effective, which can identify the polymer composition of microplastics in a wide size range.

However, challenges were encountered in the sampling of microplastics in Arctic fjords. The commonly used trawl sampling method may greatly underestimate the exposure level of microplastics in Arctic fjords.

Further studies are needed to identify the occurrence, sources, and sinks of microplastics in different water layers of the Arctic fjords and to explore the risk of exposure against microplastics.

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Author contribution All authors contributed to the study conception and design. Mengrong Bao: investigation, data curation, visualization, writing—original draft; Qinghui Huang: conceptualization; methodology; investigation; writing—original draft; writing—review and editing; Zhibo Lu: investigation design, methodology, funding acquisition; France Collard: data curation, methodology, writing—review and editing; Minggang Cai: writing—review and editing; Peng Huang: investigation; Yong Yu: project administration; Shuiping Cheng: supervision; Lihui An: instrumental analysis; Anette Wold: investigation, writing—review and editing; Geir Wing Gabrielsen: writing—review and editing; funding acquisition.

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Data Availability All data generated or analyzed during this study are included in this published article (and its supplementary information files) and available from the corresponding author on reasonable request.

Declarations

Ethical approval All ethical standards have been followed during this research.

Consent to participate Not applicable.

Consent to publish Not applicable.

Competing interests The authors declare no competing interests.

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