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Microplastics in juvenile Chinook salmon and their nearshore environments on the east coast of Vancouver Island☆

Brenna Collicutt [a, b, c, *], Francis Juanes [a], Sarah E. Dudas [a, b, c]

[a] Department of Biology, University of Victoria, 3800 Finnerty Rd, Victoria, British Columbia, V8P 5C2, Canada
[b] Department of Biology, Vancouver Island University, 900 Fifth St, Nanaimo, British Columbia, V9R 5S5, Canada
[c] Hakai Institute, 1713 Hyacinthe Bay Rd, Quadra Island, British Columbia, V0P 1H0, Canada

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ABSTRACT

Microplastics are a significant issue in the world's oceans. These small plastic particles (<5 mm in size) are becoming globally ubiquitous in the marine environment and are ingested by various fish species. Here we investigate the incidence of microplastics in juvenile Chinook salmon and their nearshore marine environments on the east coast of Vancouver Island, British Columbia. We completed a series of beach seines, plankton tows and sediment cores in nearshore areas of importance to juvenile salmon. Microplastics were extracted from fish, water and sediment samples and concentrations were quantified. Microplastics analysis, consisting predominantly of fibrous plastics, showed juvenile Chinook salmon contained 1.2 ± 1.4 (SD) microplastics per individual while water and sediment samples had 659.9 ± 520.9 microplastics m^{-3} and 60.2 ± 63.4 microplastics kg^{-1} dry weight, respectively. We found no differences in microplastic concentrations in juvenile Chinook and water samples among sites but observed significantly higher concentrations in sediment at the Deep Bay site compared to Nanaimo and Cowichan Bay sites. Chinook microplastic concentrations were relatively low compared to literature values and, given the size and type of microplastics we observed, are unlikely to represent an immediate threat to fish in this area. However, microplastics less than 100 μm in size were not included in the study and may represent a greater threat due to their ability to translocate through tissues.

1. Introduction

Juvenile Chinook salmon (*Oncorhynchus tshawytscha*) face many challenges upon entering the marine environment including change in diet, predators, and physiology associated with the adjustment to higher salinity water (Healey, 1980; Simenstad et al., 1982; Thorpe, 1994). Also during this time, juvenile salmon are met with a number of anthropogenic stressors including modified habitats (e.g., seawalls, breakwaters, marinas) and pollution (Bulleri and Chapman, 2010; Jackson et al., 2001; Lotze et al., 2006). Plastic pollution specifically has been identified as a substantial threat to marine ecosystems (Derraik, 2002; Kennish, 2002; Wright et al., 2013) including both macro (>5 mm) and microplastics (<5 mm) (Andrady, 2011). While we know more about other types of marine pollution, less is known about the threat of microplastics.

Microplastics can be produced from primary or secondary sources. Primary sources include plastics produced at the micro scale such as those in cosmetics (Fendall and Sewell, 2009) and textiles (Browne et al., 2011). In addition, secondary sources include macroplastics from litter and industry materials (e.g., derelict fishing gear) that can break down into smaller particles and eventually into microplastics (Arthur et al., 2014). Both primary and secondary sources can be transported to the marine environment through wastewater outfall (Carr et al., 2016). Microplastics have been found in most ecosystems spanning the globe including remote areas such as Arctic ice (Zarfl and Matthies, 2010) and deep-sea sediments (Van Cauwenberghe et al., 2013). The ingestion of microplastic particles has been documented in a variety of marine species ranging from zooplankton to fish to marine mammals with potential risks including internal physical (e.g., abrasion or blockage) and/or chemical damage (Cole et al., 2011; Wright et al., 2013). Microplastics can have concentrations of persistent organic pollutants (POPs) several orders of magnitude higher than ambient marine waters (Mato et al., 2001) and thus ingestion by lower

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* Corresponding author. 5865 Tweedsmuir Crescent, Nanaimo, BC, V9T5Y6, Canada.
E-mail address: Brenna.Collicutt@hakai.org (B. Collicutt).

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trophic level organisms can provide an entry point for POPs to amplify through the food chain (Teuten et al., 2009). Microplastics, and negative impacts associated with their consumption, may pose a risk to humans (Choy and Drazen, 2013; Rochman et al., 2015; Van Cauwenberghe and Janssen, 2014; Vethaak and Leslie, 2016), therefore, marine organisms essential for human food security (e.g., fish and bivalves) are a priority in microplastics research (GESAMP, 2015).

The incidence of microplastics has been documented globally in a large range of fish species (reviewed in Lusher, 2015). Because microplastics are found in both water and sediment (GESAMP, 2015), both benthic and pelagic feeders have the potential to ingest microplastics through direct consumption or by ingesting prey containing microplastics (Desforges et al., 2015; Setälä et al., 2014). Studies have yet to look at microplastics in juvenile Pacific salmon but have observed polystyrene particles in Atlantic salmon (*Salmo salar*) skin (Tang et al., 2015) and documented variable plastic abundances in many other fish species (reviewed by Lusher, 2015). Despite the number of studies that have found microplastics in fish, relatively few have examined how/if the presence of these particles impacts organismal health (Granby et al., 2018; Mazurais et al., 2015; Pedà et al., 2016; Rainieri et al., 2018; Rochman et al., 2013).

Coastal marine ecosystems have higher concentrations of microplastic pollution compared to their offshore counterparts (Desforges et al., 2014) and a large source of plastic pollution is believed to originate from anthropogenic land-based operations (Barnes et al., 2009; Browne et al., 2010; Doyle et al., 2011), therefore, nearshore environments, and the organisms that reside there, may be among the most directly and highly affected. Juvenile Chinook salmon depend on nearshore areas extensively in order to feed and grow (Healey, 1980; Levy and Northcote, 1982; Thorpe, 1994) and thus early marine residency and any significant factors affecting survival success during this time must be examined carefully.

The objectives of this study were to determine 1) the microplastic concentrations in juvenile Chinook salmon across several nearshore areas along the east coast of Vancouver Island, 2) environmental (water and sediment) microplastic concentrations, and 3) the relationships between these environmental and organismal concentrations. Project results will indicate whether there is the potential for microplastics to pose a significant threat to juvenile salmon on the east coast of Vancouver Island at this time.

2. Methods

2.1. Study site

This project took place along the east coast of Vancouver Island bordering the Strait of Georgia (Fig. 1). Sites near freshwater tributaries containing prominent Chinook salmon runs were chosen to increase capture probability as juveniles migrated to the ocean. We chose four locations including Deep Bay, Big Qualicum, Nanaimo River estuary, and Cowichan Bay. The Deep Bay site was located in the southern portion of Baynes Sound on an active shellfish aquaculture tenure consisting of both intertidal and deep-water culture. The site was in a sheltered bay with low wave exposure and slope. The Big Qualicum River is located 17 km north of Parksville and connects Horne Lake to the Strait of Georgia. This site had low slope and high wave exposure. The Nanaimo River estuary consists of an extensive tidal mud flat and our site had high wave exposure and a steep slope relative to the Nanaimo and Big Qualicum sites. Cowichan Bay receives inputs from both the Cowichan and Koksilah Rivers and is one of the biggest estuaries in British Columbia (BC) (Argue et al., 1986). Similar to the Nanaimo River site, this site had high wave exposure and a steeper slope.

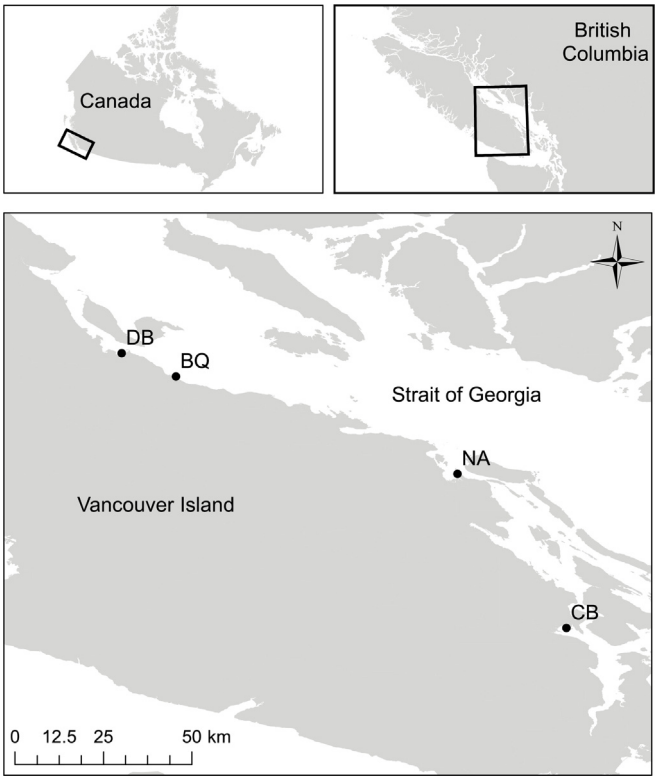

Fig. 1. Study sites along the east coast of Vancouver Island where juvenile salmon, water and sediment were sampled for microplastics presence. DB = Deep Bay, BQ = Big Qualicum, NA = Nanaimo, CB = Cowichan Bay.

2.2. Field methods

We used a 15 × 2 m beach seine consisting of 6 mm stretch mesh to capture juvenile Chinook salmon during June and July 2015 at each site. Beach seines were set at low tide windows between 0.5 and 1.1 m for Deep Bay, Big Qualicum and Cowichan Bay sites. The Nanaimo River site was an exception because it was accessible at high tides only, therefore sampling was conducted during 2.8–3.7 m tides. Between 3 and 8 beach seine sets were completed at each site until at least 40 fish were captured or catch per seine was below one.

The beach seine was set using an outboard aluminum boat. Captured fish were corralled into the beach seine bunt and placed into a 1.2 × 0.6 m floating PVC rectangle. This allowed quick sorting, identification and release of non-target species without removing them from their natural environment. Juvenile Chinook were identified using characteristics in Hartman (1997), enumerated and euthanized (up to a maximum of 40 per site) using an overdose of MS-222 (300 mg/L) and stored on ice.

We collected five 25 L surface water samples during low tide at each site spanning approximately 100 m (~25 m between samples) of shoreline where fish sampling took place. We filtered the water samples using a 100 μm plankton net with a 30 cm diameter opening. Contents were rinsed and concentrated into the codend then transferred into sample containers. GPS locations were recorded and samples were stored at −12 °C.

Sediment samples were taken in the mid tidal zone (1.5–2.2 m) using a PVC sediment corer. Eight samples of 15 cm depth by 4 cm diameter were taken approximately 1 m apart and stored in Ziploc bags. GPS locations were recorded and samples were stored at −12 °C.

2.3. Lab methods

Upon return to the laboratory, fish were scanned for pit tags and their identification was confirmed using external features such as parr marks and anal fin shapes, in addition to branchiostegal and pyloric caeca counts (Hartman, 1997). Fish were measured for fork length (±1 mm) and weighed (±0.01 g). Stomachs were removed by making an incision from the anus to the operculum along the ventral surface of the fish, pulling out the intestine from the anus, clipping the esophagus, and removing the entire gut. The gut was stored in a 20 mL glass scintillation vial containing 70% ethanol until further processing. During microplastic extraction, scintillation vials containing fish guts were covered with 100 μm nylon mesh and dried at 60 °C overnight (or until sample was completely dry) in a conventional laboratory drying oven. Once dry, samples were crushed using the blunt end of a stainless-steel probe and 10–15 mL of 10% filtered potassium hydroxide (KOH) was added to the vial to digest organic material. Samples were covered with foil, returned to the drying oven and allowed to digest for 24 h at 60 °C. Following organic matter digestion by KOH, samples were vacuum filtered over a Whatman GF/C 1.2 μm glass microfiber filter paper. Resulting filter papers were stored in sealed petri dishes until microscopic examination.

Water samples were subsampled in 50 mL portions, placed into 500 mL glass mason jars, and covered with 100 μm mesh secured by the mason jar screw top. Moisture was removed through the mesh by inverting the mason jar over a filter flask attached to a vacuum pump. Three times the sample volume (~100–200 mL) of KOH was added to each mason jar and placed in the drying oven for 24 h at 60 °C to digest organic material. After 24 h, KOH was removed with the vacuum filter (as above) and the sample was rinsed with filtered deionized water (FDI) to remove any excess KOH. Samples were centrifuged with FDI at 3500 g for 10 min in 50 mL centrifuge tubes and supernatant was poured off into a FDI rinsed flask. Samples were then centrifuged with filtered saturated calcium chloride ($CaCl_2$: density: 1.38–1.40 g mL^{-1}). This process was repeated a total of three times, pouring off supernatant and shaking vigorously to break apart the concentrated sample pellet each time. Combined supernatants were then vacuum filtered over Whatman GF/C 1.2 μm glass microfiber filter paper. Resulting filter papers were stored in sealed petri dishes until microscopic examination.

Sediment samples were dried in tin foil covered foil pans for 48–72 h in a conventional drying oven at 70°C. Samples were homogenized using a ceramic pestle and approximately 70–80 g of sediment was sieved through a 4.75 mm and 90 μm mesh, 7.5 cm diameter stainless steel sieve set. Sediment grains larger than 4.75 mm and smaller than 90 μm were discarded. Sediment was transferred to a 500 mL glass mason jar and covered with 100 μm mesh secured by a mason jar screw top. Excess moisture was removed through the mesh by inverting the mason jar over a filter flask attached to a vacuum pump and samples were dried for 24–48 h. Dried sediment weight was determined indirectly by weighing the mason jar and dried sediment then weighing the empty jar following the extraction process. Approximately 100–200 mL of 10% filtered KOH was added to the mason jars containing the dried sediment and placed in the drying oven at 60°C for 24 h to remove organic matter. KOH was removed from samples using the vacuum pump as above and rinsed three times with FDI. Sediment was partitioned into five 50 mL centrifuge tubes in approximately 10 g increments. Samples were then centrifuged and filtered as per water samples.

The process of microplastic enumeration was identical for all sample types. Filter papers with samples were placed on a FDI dampened gridded glass petri dish and examined under a dissecting microscope. Using 40–50x magnification, filter papers were systematically examined for microplastic particles. Under this magnification, characteristics such as bendability, colour and twistedness were used for initial microplastic assessment. Potential microplastics were mounted on glass slides and examined under a compound microscope. At the higher magnification, characteristics such as structure and colour were used to further assess potential microplastics particles. Microplastics were enumerated and classified by colour and type (fibre, fragment, film or pellet). The codend we used to filter water had a 100 μm mesh, therefore, this was deemed the lower size limit of plastics in water, fish and sediment samples.

2.4. Contamination control

Several steps were taken to eliminate plastic contamination of our samples. Any solution used during microplastic extraction was made using deionized water and filtered over a Whatman GF/C 1.2 μm glass microfiber filter paper. Non-plastic equipment was used as much as possible and all glassware and equipment was washed thoroughly with soap and water followed by a FDI wash consisting of three separate rinses. Immediately following FDI rinses, glassware and equipment were covered with tin foil to protect from airborne plastic contamination. Equipment such as forceps were rinsed with FDI immediately before each use and samples were kept covered as much as possible throughout processing. When transferring samples between different vessels, thorough rinses (x3) were done to ensure complete transfer of material. All work was completed in a laminar flow hood and laboratory workers wore natural fibre clothing and 100% cotton laboratory coats in the laboratory at all times during microplastic extraction processing. At least three procedural blanks were run in conjunction with each sample batch to determine the amount and types of contamination present in our samples. Plastic contamination determined from procedural blanks was averaged by colour and subtracted from each sample in the corresponding batch. All results presented were corrected in this manner for contamination.

2.5. Statistics

Microplastic concentrations were compared among sites using a one-way analysis of variance (ANOVA). In the case of water and sediment samples, microplastics concentrations were square root transformed to meet normality assumptions. When differences existed among sites, a Tukey post-hoc test was used to assess which sites were significantly different. Fish microplastics data were not normal after transformation and so a Kruskal-Wallis rank sum test was performed to determine differences among sites. Spearman's rank correlation was used to determine relationships between juvenile Chinook and environmental (water and sediment) microplastic concentration levels. The number of plastic particles in fish was converted to microplastics g^{-1} to standardize across fish sizes while plastics in water samples were converted to microplastics m^{-3} and sediment plastics were expressed as microplastics kg^{-1} dry weight (d.w.).

3. Results

In all sample types the majority (>90%) of microplastics were fibrous in nature as opposed to fragments, films or pellets. The remaining microplastics identified were fragments and, although field contamination samples were not taken specifically, all fragments were suspected to be originating from our equipment (e.g., plankton net and falcon tube lids). Therefore, all results will refer to fibres only. All plastics found were 100–5000 μm in length and

approximately 10–20 μm in diameter. All values are presented ± standard deviation.

We extracted microplastics from 74 juvenile Chinook salmon (fork length: 78.2 ± 9.3 mm SD; wet weight: 6.01 ± 2.33 g) across four sites along the east coast of Vancouver Island including Deep Bay ($n = 19$), Big Qualicum ($n = 9$), Nanaimo ($n = 7$) and Cowichan Bay ($n = 39$). Of the juvenile salmon sampled, 59% contained at least one plastic particle with a mean of 1.15 ± 1.41 microplastics per individual (0.24 ± 0.36 microplastics g^{-1}) across all sites. Fibrous microplastics made up 95% of plastic particles identified in juvenile Chinook salmon. There were no significant differences among the number of microplastics g^{-1} of juvenile Chinook sampled across our four sample sites (Kruskal-Wallis, $p = 0.490$; Fig. 2A). The most abundant fibres by colour were clear (41%) and blue (20%) for all sites.

The mean microplastic concentration observed over all water samples was of 659.88 ± 520.87 m^{-3} with 100% of the samples containing at least one fibrous microplastic particle. As in fish, the majority of plastic particles found (93%) were fibrous plastics. There were no significant differences in the average number of plastic fibres m^{-3} in water samples across our four sample sites (ANOVA, $p = 0.571$; Fig. 2B). Similar to the juvenile Chinook samples, clear (70%) and blue (12%) fibres were the most abundant across all sites.

The mean microplastic concentration in sediment samples was 60.2 ± 63.4 microplastics kg^{-1} d.w. with 87.5% of samples containing at least one microplastic particle. The majority (91%) of microplastic particles identified were fibres. We found significant differences in microplastic concentrations among sites (ANOVA, $p = 0.015$; Fig. 2C). Deep Bay had significantly more microplastics than Nanaimo (Tukey post-hoc, $p = 0.020$) and Cowichan Bay (Tukey post-hoc, $p = 0.045$), while Big Qualicum was not significantly different from any other site (Tukey post-hoc, $p > 0.05$). By colour, clear was the most frequently identified microplastic fibre (64%) while blue (15%) was the next most common at all sites.

There were no significant relationships between juvenile Chinook and environmental microplastic concentrations ($p > 0.05$) nor was there a significant relationship between the environmental (water and sediment) levels ($p > 0.05$).

4. Discussion

Microplastics may be a significant threat to marine ecosystems (Andrady, 2011) and juvenile Chinook salmon may be particularly at risk during a critical time in their lifecycle. In this study, juvenile Chinook salmon, water and sediment samples were examined to determine microplastic abundances. Relative to other studies of microplastics in fish (Jovanović, 2017), low concentrations of plastic fibres in fish samples were observed which were not correlated with water and sediment microplastic concentrations.

We sampled 74 juvenile Chinook across four sites and found that 59% contained at least one plastic fibre. Although previous studies have shown relatively high variance in the proportion of fish containing microplastic particles (range: 0.3%–77%; summarized in Jovanović, 2017) our results suggest a relatively high proportion of fish with at least one plastic particle in their gastrointestinal tracts.

The consequences of microplastic ingestion are poorly understood and the extent and type of impact appears to depend on exposure concentration, plastic type, shape and size, as well as associated chemicals. In addition, responses differ among species and size (Wright et al., 2013) with early life stages potentially being more vulnerable (Mazurais et al., 2015). Effects on fish include structural intestinal alteration with potential implications for nutrient uptake (Pedà et al., 2016), liver toxicity (Rochman et al., 2013) and associated increased mortality (Mazurais et al., 2015; Pedà et al., 2016; Rochman et al., 2013). Mazurais et al. (2015) and Pedà et al. (2016) both conducted laboratory studies with European sea bass (*Dicentrarchus labrax)* and each tested a different type of plastic pellet (polyethylene and polyvinylchloride,

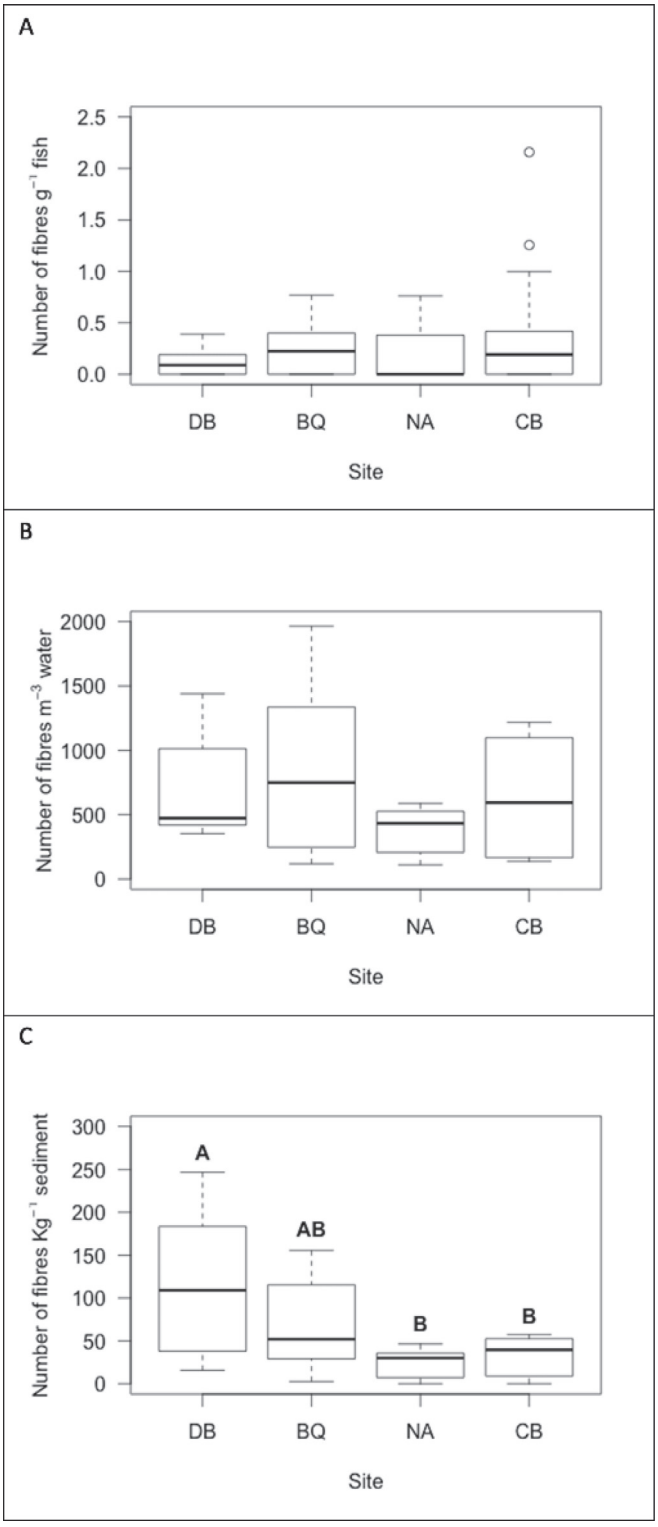

Fig. 2. Average number of plastic fibres g^{-1} of fish (A), fibres m^{-3} of water (B) and fibres Kg^{-1} of sediment (C) across samples sites including Deep Bay (DB), Big Qualicum (BQ), Nanaimo (NA) and Cowichan Bay (CB). Boxes represent interquartile ranges with medians (black line) and the whiskers are minimum and maximum values. Open circles represent outliers and different letters represent significant differences among sites.

respectively). Both studies found significant health consequences (e.g., intestinal alterations, mortality), however, the type, extent and severity varied substantially. In addition to differences in plastic characteristics, studies examining microplastic ingestion in fish also look at a wide variety of fish species and life stages. Lu et al. (2016) determined that Zebrafish (*Danio rerio*) exposed to polystyrene demonstrated liver inflammation and lipid accumulation, oxidative stress and metabolic alterations. Laboratory studies of the common goby (*Pomatoschistus microps*) demonstrated reduced predatory performance and efficiency when exposed to polyethylene microspheres (de Sá et al., 2015). If juvenile salmon experience similar impacts, their fitness and survival could be affected as growth is critical during this early marine life stage (Beamish and Mahnken, 2001).

The average microplastic concentration per individual fish in this study (1.15 ± 1.41) is similar to other average concentrations observed previously ranging from 0.01 to 3.8 particles/fish (Jovanović, 2017 and references therein). In a study of microplastic particle ingestion by zooplankton in the northeast Pacific Ocean, Desforges et al. (2015) estimated that juvenile salmon in coastal BC could consume 2–7 microplastic particles per day. Mazurais et al. (2015) observed that a high proportion of polyethylene beads were being egested by sea bass larvae within 48 h therefore other species such as salmon may do the same. If so, our microplastic quantities observed in our juvenile Chinook are likely underestimations of total microplastic ingestion. In addition, microplastics smaller than 100 μm were not included in this study leading to further underestimations.

Despite high egestion rates, Mazurais et al. (2015) determined that a plastic loading of 3.3 ± 0.19 polyethylene beads in larval European sea bass caused significantly higher mortality than the control group. Given the size difference in fish between the sea bass study (~2–12 mg) and the current study (6010 ± 2330 mg), we could predict that the same plastic loadings would have a less severe impact on our juvenile Chinook and, although species-specific responses may differ between sea bass and salmonids, it is likely that the average microplastic concentration of 1.15 ± 1.41 found in our study is not causing significant mortality events.

Across all water samples, we found a mean of 659.88 ± 520.87 microplastics m^{-3}. Desforges et al. (2014) estimated a mean of 3210 ± 628 microplastics m^{-3} in the Strait of Georgia which is higher than those found in this study, however, differences in sampling methodologies (i.e., looking at a larger size range: 62.5 μm–5 mm) may partially account for their higher estimates. Desforges et al. (2014) also found that plastic particle size increased with distance from shore, indicating that the smaller size fractions may be more abundant in nearshore coastal sites like ours. Although our concentrations were low compared to Desforges et al. (2014), we found relatively high concentrations compared to studies from other coastal regions worldwide that, for the most part, observed <1 microplastic m^{-3} (reviewed in Lusher, 2015). As most studies used a mesh size of 333 μm (GESAMP, 2015), however, results are likely underestimations for reasons outlined above. These findings emphasize a need for standardization and the use of smaller mesh sizes to accurately assess microplastic concentrations worldwide.

Across all sites, we found a mean of 60.2 ± 63.4 microplastics kg^{-1} d.w. of sediment sampled. Cluzard et al. (2015) sampled sediment from various sites throughout Baynes Sound and found the median number of microplastics kg^{-1} d.w. to range from 0 to 85. In the region that included a sample area in a similar location to our Deep Bay site, a median of 24.6 microplastics kg^{-1} d.w. was documented which was lower than the mean of 115.40 microplastics kg^{-1} d.w. found in our study at that site. This inconsistency may be due to differences in plastic extraction techniques (i.e., NaCl flotation) which may lead to underestimations of higher density plastics in Cluzard et al. (2015). In addition, their study only looked at plastic particles larger than 1 mm in size which would also account for the higher concentrations found in our study as we included plastics from 100 μm to 5 mm in size. Microplastic concentrations in Cluzard et al. (2015) and our study were also consistent with many European coastal studies (Browne et al., 2011; Claessens et al., 2011; Thompson et al., 2004) but lower than Mathalon and Hill (2014) who documented microplastic ranges of 200–800 microplastics kg^{-1} in sediment from Halifax Harbour, Nova Scotia. Mathalon and Hill (2014), however, did not correct their values despite high laboratory contamination levels and therefore, microplastic concentrations are likely overestimations. Sediment microplastics loads will also be affected by plastic type (e.g., PVC is a higher density plastic that will tend to sink in lower density salt water more readily than lower density plastics such as polyethylene (Van Cauwenberghe et al., 2015)), geography and oceanography (Vianello et al., 2013), sediment type (Strand et al., 2013), and amount of environmental biofouling (Barnes et al., 2009; Browne et al., 2010; Thompson et al., 2004).

Among sites, we only found differences in sediment microplastic concentrations. Because fish, water and sediment move throughout ecosystems, microplastics may disperse far from their original source, however, microplastics in sediments may be retained longer due to their ability to form aggregates with organic matter (Clark et al., 2016; Long et al., 2017; Summers et al., 2018) which may cause a faster rate of sinking from the water column and higher accumulation on the ocean floor. In other words, water and fish samples may represent a more transient microplastic loading while sediment may potentially offer a longer time scale in which to study. (Davis and Murphy, 2015).

Significantly higher microplastic abundances were observed in sediments at Deep Bay compared to the Nanaimo and Cowichan sites. The Deep Bay site is in a sheltered bay and has a relatively shallow slope while both the Nanaimo and Cowichan Bay sites are in more exposed areas with steeper slopes. Various studies suggest that exposed areas with higher wave energy may accumulate less microplastics and the enclosed geometry of harbours, for example, may trap microplastics and increase benthic accumulation (Claessens et al., 2011; Cluzard et al., 2015; Vianello et al., 2013). Therefore, differences in site morphology may result in higher microplastic retention in sites like Deep Bay where plastic would likely settle more readily. Claessens et al. (2011) speculated that harbour activities (e.g., recreational boating) added to the microplastic load which is also consistent with our study as the Deep Bay site is within close proximity to the Deep Bay Marina. Furthermore, sites with freshwater inputs generally have higher microplastics loads (Claessens et al., 2011; Vianello et al., 2013) and although all of our sites had freshwater inputs, the Deep Bay and Big Qualicum sites, which had the highest sediment microplastic concentrations, were in the closest proximity to freshwater tributaries with Chef Creek and Big Qualicum River, respectively, emptying directly into the sites. The Nanaimo and Cowichan Rivers may also influence microplastic loads in their associated sites but to a lesser extent as they were further away (~2 km).

In addition, the Deep Bay sample site is located on a shellfish aquaculture tenure which may have direct plastic inputs through the use of plastic netting and ropes. Although the other sites have a range of other potential plastic sources, none are in as close proximity to a source as the Deep Bay site. Aquaculture has been

identified as a potential microplastics source (Cole et al., 2011; Desforges et al., 2014; Hinojosa and Thiel, 2009; Mathalon and Hill, 2014) and cannot be discounted, however, these higher concentrations may also be linked to proximity to the harbour, freshwater, oceanography and bathymetry.

We found no significant correlations between the amount of microplastics found in water or sediment samples with the amount ingested by juvenile Chinook indicating that environmental levels do not necessarily reflect the amount ingested by marine organisms in this area. We also found that water microplastic concentrations do not correlate with sediment concentrations from the same sites. This may reflect differences in low density (floating) and high density (sinking) plastics across each site or differential biofouling that may be causing low density plastics, that would normally float, to sink (Barnes et al., 2009; Browne et al., 2010; Thompson et al., 2004). Additionally, plastic sinking rates may be altered through ingestion by zooplankton followed by egestion via fecal pellets (Cole et al., 2016). This disparity among water, sediment, and fish samples may also be a product of the patchy distribution of plastics in the marine environment (Moreira et al., 2015) demonstrated by the relatively high standard deviations across all three types of samples.

Microplastics can originate from several different sources and through a combination of oceanography, wind and marine organisms, the transfer and dispersal of plastic particles can be extensive. Because of this complexity, linking microplastics to a specific source is challenging. Deductions can be made, however, based on various plastic characteristics. For example, we found that the majority of microplastics found in juvenile Chinook, water and sediment samples were fibrous in nature as opposed to being a film, fragment or pellet. Desforges et al. (2014) found a negative relationship between the proportion of fibres in the plastic samples and distance from shore indicating that nearshore areas have generally higher fibre content than offshore areas. Moreover, they estimated that samples from areas we examined would have greater than 80% fibre content which is consistent with our findings. A large source of fibrous microplastics originate from textiles and through sewage output with up to 1900 fibres per garment per wash being released from household washing machines (Browne et al., 2011). Given the dominance of fibrous plastics, sources of microplastics found in this study may include textiles in addition to synthetic fishing line, nets and rope used in fishing and aquaculture industries (Claessens et al., 2011).

Given the rapid advancement of the microplastics field since this study was conducted, we recognize that our method of visual counting without the use of further equipment and chemical analysis (e.g., Fourier transform infrared spectroscopy, Raman spectroscopy, pyrolysis gas chromatography with mass spectrometry) may have led to microplastic overestimations, however, relative media and site comparisons in this area remain interesting and novel. Future work would benefit from using visual counting in combination with further analytical methods for more standardized results (Lusher et al., 2017; Mai et al., 2018; Song et al., 2015).

In conclusion, we examined the incidence of microplastic particles in juvenile Chinook and their nearshore environment along the east coast of Vancouver Island. We found predominantly microplastic fibres in the majority of our samples supporting the notion that microplastics are widespread and ubiquitous in the marine environment. We did not find significant differences in microplastic abundances ingested by juvenile Chinook or in water samples among our sample sites but found significantly more plastic fibres in sediment at our Deep Bay site. This difference may be due to geographical differences in oceanography and potential source inputs (e.g., shellfish aquaculture, marina, recreational boating) in the surrounding area. Our results demonstrate that environmental concentrations (i.e., water and sediment) do not necessarily reflect the amount of plastics found in juvenile Chinook salmon. These results also show that plastic concentrations vary within habitat types in the environment likely because sediment may gather higher microplastic abundances due to aggregation and accumulation at these sites.

Our microplastic abundances in salmon were low based on existing knowledge and seem unlikely to cause negative impacts, however, further studies on plastic retention time, smaller plastic size classes and health implications in juvenile Pacific salmon are necessary to confirm this. Studies examining the ingestion pathway (direct vs. indirect) and bioaccumulation at environmentally relevant concentrations would also benefit this field of study by improving environmental and organismal microplastic predictability. Microplastic fragments in surface water continues to rise as global plastic production increases (Barnes et al., 2009) indicating that although microplastic particles may not be an immediate threat to juvenile Chinook salmon, the future is uncertain. As there is little information on bioaccumulation of plastics and their pollutants, it is unclear how whole ecosystems may be affected and what the implications are for human health (Vethaak and Leslie, 2016).

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