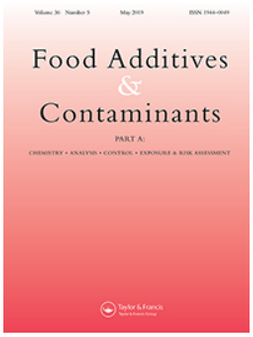

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Microplastics in coastal areas and seafood: implications for food safety

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ABSTRACT

Microplastics have become ubiquitous in the marine environment. Microplastics have been detected in many coastal environments and species, including commercial seafood. This triggers concern about potential economic impacts and the risks of dietary exposure, especially for coastal communities. However, data regarding the levels of microplastics in coastal seafood and their toxicological effects are still limited. Accordingly, the dietary risk is still poorly explored. This review summarizes and discusses recent scientific findings on (i) the presence of microplastics in coastal waters, (ii) the occurrence of microplastics in coastal seafood and the likelihood of trophic transfer, and (iii) the effects of microplastics on coastal fish and shellfish species. Human toxicity data are also reviewed, but the risks for human health are difficult to determine due to limited data. Based on available worldwide data, the estimation of microplastics intake through seafood consumption shows a huge variation. Additionally, a lack of standardized analytical methods complicates the comparison of results between studies and therefore seriously affects the reliability of risk assessments. It is concluded that more exposure and toxicity data are needed properly to assess human health risks of microplastics in coastal seafood, and the lack of data currently impede the derivation of a risk-based food safety standard. The pros and cons of an interim solution, i.e. setting a provisional action level, are being discussed.

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Microplastics; coastal seafood species; trophic transfer; food safety

Introduction

After the introduction of plastics in the 1950s, their intensive use and poor waste management have led to a widespread dispersal of plastics in the marine environment (Jambeck et al. 2015). The growing presence of plastic pollution has triggered serious concerns about the implications for marine ecosystems, the ecosystem services provided, and human health (GESAMP 2016). The pollution expands from shorelines to the open ocean and the deep seas (Bouwmeester et al. 2015; GESAMP 2016).

It has been estimated that 4.8–12.7 million tons of plastic waste ended up in the ocean in 2010 (Jambeck et al. 2015). The broad range in plastic marine debris estimations is caused by the variation in population size and the quality of waste management systems in the 192 coastal countries targeted in the study (Jambeck et al. 2015). The pollution originates from both terrestrial and aquatic sources. The litter from terrestrial sources usually originates from urban areas, tourism and river outflows, whereas marine debris originates from ships or abandoned, lost, or otherwise discarded fishing gear, and will typically be deposited along the shore when entrapped in near-shore currents (Ryan et al. 2009). Another study estimated that between 1.15–2.41 million tonnes of plastic waste enters the ocean every year from rivers (Lebreton et al. 2017).

Although plastics are generally persistent and durable, photo-oxidative degradation caused by prolonged exposure to ultraviolet (UV) radiation and physical abrasion can fragment plastic debris into smaller particles in the micrometer to nanometer range (Andrady 2011). Although there is no generally accepted definition of microplastics, they are commonly referred to as plastic particles with a size smaller than 5 mm (Law and Thompson 2014). In this review, we consider microplastics to be in the size range

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Supplemental data for this article can be accessed on the publisher's website.

1 µm to 5 mm as proposed by Frias and Nash (2019).

Distinction is made between primary and secondary sources of microplastics (Cole et al. 2011; GESAMP 2016; Boucher and Friot 2017). Primary sources are direct emissions of 'primary' microplastics by industrial processes and domestic uses. Primary microplastics are manufactured in a micro-size range, such as pellets, powder plastics (GESAMP 2016) and microbeads in cosmetics (Cole et al. 2011; GESAMP 2016). Primary microplastics also can be derived from the abrasion of large plastic objects during manufacturing, use or maintenance, e.g. the microfibres resulting from the abrasion of synthetic textile during washing (Boucher and Friot 2017; Carr 2017). Secondary sources of microplastics are the result of fragmentation and degradation of macroplastics exposed to UV and/or physical abrasion in the environment (GESAMP 2016; Boucher and Friot 2017).

Microplastics show a huge variation in physical and chemical characteristics such as size, composition, weight, shape and colour (Hidalgo-Ruz et al. 2012). These characteristics influence the behaviour of the particles, e.g. their dispersion in the marine environment, adsorption and absorption of contaminants, microbial colonization, potential bioavailability and toxicity (Lambert et al. 2017; Potthoff et al. 2017). For example, low density particles floating on the surface are more prone to advective transport than sinking high density particles. Likewise, spheres may be more likely to be absorbed by organisms than fibres (Au et al. 2015), while irregularly shaped microplastics can sorb less persistent organic pollutants such as phenanthrene, but have a longer gut residence time in *Daphnia magna* (Frydkjær et al. 2017). Furthermore, irregularly shaped microplastics are likely to cause different physiological and biochemical responses in fish compared to spherical microplastics (Choi et al. 2018).

Several studies have reported a high abundance of microplastics in densely populated coastal areas (Lebreton et al. 2017; Ling et al. 2017). Coastal areas are typically located close to densely populated areas and human activities (Browne et al. 2011; Reisser et al. 2013).

The presence of microplastics near the coast is influenced by physical and chemical processes, including the transport of plastics by current and wind, as well as degradation by weathering (Fok et al. 2017; Hartmann et al. 2017; Zhang 2017; Yu et al. 2018). Some studies revealed that sandy beaches may act as a temporary or permanent sink for microplastics (Lozoya et al. 2016; Yu et al. 2016). Plastics in beach sediments generally have a long residence time and can be fragmented due to UV irradiation and physical abrasion by waves (Veerasingam et al. 2016). In the ocean, fragmentation is much slower than on the beach because of the lower temperatures, UV intensity and mechanical abrasion (Andrady 2011; Cole et al. 2011).

Coastal areas are an important source of seafood. The harvest of coastal fishes reached 7.5 million tonnes in 2014 (excluding crustaceans and molluscs), constituting approximately 8% of the global marine harvest of fish, crustaceans and molluscs (i.e. 93.4 million tonnes) (FAO 2016a, 2016b). Several exotic and expensive seafood species are cultured in coastal areas, e.g. shrimps and crabs. Based on FAO data, marine and coastal aquaculture produced about 28 million tonnes in 2015, which includes molluscs (65%), crustaceans (28%) and finfish (7%) (FAO 2016b). Oysters, clams and mussels are the main molluscan species, whereas penaeid shrimps are the most important crustaceans (Romeo et al. 2015). These amounts underline the economic importance of coastal fishery and aquaculture.

Microplastics can be transferred from the environment to organisms, and subsequently pass through the food web, i.e. transfer from prey to predator. Ingestion of microplastics has been documented in many seafood species from various places around the world, including fish (Foekema et al. 2013; Lusher et al. 2013; Neves et al. 2015; Rochman et al. 2015; Brate et al. 2016; Naidoo et al. 2016; Tanaka and Takada 2016; Jabeen et al. 2017), bivalves (De Witte et al. 2014; Van Cauwenberghe and Janssen 2014; Rochman et al. 2015; Davidson and Dudas 2016; Santana et al. 2016; Leslie et al. 2017), molluscs and crustaceans (Devriese et al. 2015; Wójcik-Fudalewska et al. 2016). Ingestion can be selective (i.e., intentional feeding on plastic fragments that resemble natural food in size and appearance) or non-selective (i.e., particles randomly ingested as a result of suspension, deposit or filter feeding behaviour) (Bellas et al. 2016; Wesch et al. 2016; Santillo et al. 2017). Several studies have shown that the ingestion of

microplastics can cause histopathological changes (Peda et al. 2016) and may affect growth (Watts et al. 2015), the reproduction system (Rochman et al. 2014), and behaviour (Mattsson et al. 2015; Tosetto et al. 2016) of marine organisms such as fish and crab. Obviously, also humans can be exposed to microplastics through the consumption of seafood. However, the relevance and implications of microplastics exposure for seafood species and human health are still largely unknown (Bouwmeester et al. 2015; Koelmans et al. 2016; Barboza et al. 2018; Smith et al. 2018).

Although many studies on microplastics in coastal areas and seafood species have been conducted, the available information is still fragmentary. The main aim of the current review is to collate this information and to present a state-of-the-art global overview on the occurrence of microplastics in coastal areas and coastal seafood species. A comprehensive search for scientific literature was conducted to identify peer-reviewed original research dealing with the presence of microplastics in coastal areas and in seafood collected from coastal areas. The boundary of the coastal areas was set at roughly 100 km distance from the shoreline (Christian et al. 2005). The data retrieved are discussed within the context of the available knowledge on the effects of microplastics on coastal fish and shellfish species, and human health. We conclude with a discussion on how to deal with microplastics as a novel food contaminant from a food safety perspective.

For the present study, we analysed literature that was available up to August 2018. A comprehensive search was conducted for peer-reviewed original research papers, technical reports and proceedings dealing with the presence of microplastics in coastal regions and in seafood collected from coastal regions. The search strategy was designed to identify scientific literature on microplastics in coastal regions and coastal seafood species, using the following search terms: microplastics, coastal, seafood, analysis, toxicity, toxicokinetics, and human exposure. Literature was retrieved from various databases, i.e. Web of Science, Science Direct, Directory of Open Access Journals (DOAJ), Wiley Online Library, Council of Australian University Librarians (CAUL) Taylor and Francis Journals, and PubMed Central. The eligibility of the literature obtained was assessed based on the relevance and novelty of the study, and the reputation of the journal (i.e. the impact factor >1). There were 323 peer-reviewed papers identified through database searching and 166 articles were selected to be used in this review. Both qualitative and quantitative information of the collected data was grouped into one of the following categories: (1) microplastics in coastal regions, (2) microplastics in coastal seafood and trophic transfer, (3) the effects of microplastics on marine species. All data including the sampling location, quality assurance implementation during microplastic analysis, the abundance or concentration of microplastics in both water and sediment samples, the number or concentration microplastics particles in seafood samples, and type and the size of plastics were collected. Since this review focuses on coastal seafood, seafood species living in other habitats than coastal areas were excluded. The obtained data on the abundance of microplastics in both sediment and water, as well as on the concentration of microplastics in seafood samples, were normalized, i.e. expressed in a common metric. For toxicity studies, the type of seafood, the type and size of microplastics, the level and duration of exposure, and the toxic effects of the microplastics were extracted and evaluated to assess the impact of microplastics on the organisms, and, where possible, on human health. All raw data extracted from literature were compiled in an Excel spreadsheets (see Supplementary Table S1–S7).

Microplastics in coastal areas

Microplastics have been analysed in sediments and waters of a wide variety of coastal areas around the globe, i.e. from very remote to densely populated areas (Tables 1 and 2) and as well as in coastal seafood (Tables 3 and 4). The data provide a first global impression of the relative spatial distribution of microplastics. The number of studies on microplastics in Asian countries has increased substantially compared to previous reviews (Barnes et al. 2009; Browne et al. 2011; Avio et al. 2016; Auta et al. 2017). All microplastics levels were normalized by expressing the concentration in similar units, but it should be kept in mind that the wide array of methods used for

Table 1. The concentration of microplastics (average or range or median in particles/kg dw) and type of plastics in sediments in coastal areas worldwide.

Location	Concentration (particles/kg dw)	Particle size	Type(s) of plastics detected	Method of microplastics identification	QA	Reference
Asia						
Singapore, coastal mangrove	12–62.7	<20 – >5000 µm	PA, PE, PP, PVC	ATR-FTIR	NR	Hazimah et al. (2014)
Malaysia, beach	8.58	1–30 mm	NR	NR	NR	Fauziah et al. (2015)
Taiwan, northern coast	54.8	0.25 – ≥4 mm	ABS, PE, PP, PS	ATR-FTIR and SR-FTIR microscopy	NR	Kunz et al. (2016)
Hong Kong, beach	50.7	0.315 – >5 mm	NR	NR	NR	Cheung et al. (2016)
China, Beibu Gulf, coastline	5,020–8,720	< 1 – <5 mm	PE, HDPE, PET, PS	FTIR microscopy	NR	Qiu et al. (2015)
China, north Bohai Sea, beach	102.9–163.3	NR	Alkyd, PE, HDPE, LDPE, PET, PEVA, PP, PS	ATR-FTIR	+	Yu et al. (2016)
South Korea, Soya Island, beach	0.9–4,463	50 – >5,000 µm	PE, PP, PS, EPS, PU	Vacuum FTIR and FTIR microscopy	NR	Kim et al. (2015)
Iran, Khark Island, coast	295–1,085	≤100 – ≥5000 µm	NR	NR	+	Akhbarizadeh et al. (2017b)
Iran, Strait of Hormuz, coastline	(1) – 1,258	>0.025 – <4.75 mm (diameter) 1.4–50 mm (length)	PA, PE, PET	FTIR	NR	Naji et al. (2016)
Europe						
Spain, Mallorca Island and Cabrera Island, coast	100.7–897.3	>0.063 – <2mm	NR	NR	+	Alomar et al. (2016)
Portugal, Algarve, coast	10	>1 µm	PP, rayon	FTIR microscopy	+	Frias et al. (2016)
Portugal, western coast, beach	28.9	<1 – >10 mm	PE, PL, PS	FTIR microscopy	NR	Martins and Sobral (2011)
Germany, Baltic Sea, coast	0–7	55 µm – 1 mm	NR	NR	+	Stolte et al. (2015)
Belgium, North Sea, beach	92.8–166.7	38 µm – 1 mm	PA, PE, PP, PS, PVA	FTIR microscopy	NR	Claessens et al. (2011)
Netherlands, North Sea, coast	100–3,600	10–5,000 µm	NR	NR	+	Leslie et al. (2017)
Slovenia, beach	177.8	0.25–5 mm	NR	NR	NR	Laglbauer et al. (2014)
Poland, Baltic Sea, coastline	25	0.1–5 mm	PL	FTIR microscopy	+	Graca et al. (2017)
	53		PE, PP, PVAc			
	0–27		PL			
North America						
Canada, eastern shore of Nova Scotia, beach	2,200–7,400	9–220 µm (median)	NR	NR	+	Mathalon and Hill (2014)
Mexico, Huatulco, beach/sediment			NR	SEM	NR	Retama et al. (2016)
(April)	0–4,800	3.0–50 µm (thickness)				
(December)	200–6,900	0.0043–4.5 mm (length)				
South America						
Brazil, Guanabara Bay, beach		NR	PA, PP, PVA, PU, styrofoam	NR	NR	De Carvalho and Neto (2016)
(during summer)	0.15–1.63					
(during winter)	0.04–9.3					
Uruguay, Punta del Este, beach	0.8	0.3 – >100 mm	PE, PET, PP, PS, PVC	Raman microscopy	NR	Lozoya et al. (2016)

ABS: acrylonitrile butadiene styrene, PA: polyamide, PE: polyethylene, HDPE: high density polyethylene, LDPE: low density polyethylene, PET: polyethylene terephthalate, PEVA: polyethylene vinyl acetate, PL: polyester, PP: polypropylene, PS: polystyrene, EPS: expanded polystyrene, PU: polyurethane, PVA: polyvinyl alcohol, PVAc: polyvinyl acetate, PVC: polyvinyl chloride, ATR-FTIR = Attenuated Total Reflection Fourier transform infrared, SR-FTIR = Synchrotron radiation-based Fourier transform infrared, NR = Not Reported, QA = Quality Assurance, + = the study implemented measures to prevent microplastics contamination such as the analysis of blank or spiked samples, cleaning of all equipment and contact materials, and wearing 100% cotton coat during analysis

Table 2. The concentration of microplastics and type of plastics in the water column of coastal areas worldwide.

Location	Concentration (particles/m^3)	Sampling depth	Particle size	Type of polymer(s)	Method of microplastics identification	QA	Reference
Asia							
Indonesia, Cilacap coast	0.27–0.54	Sea surface (20 cm)	< 2.5 – > 5 mm	PET, HDPE, PVC, LDPE, PP, PS, PC	ATR-FTIR	NR	Syakti et al. (2017)
Japan, Seto Inland Sea	0.4	Sea surface (75 cm)	0.1 – >10 mm	PE, PP	FTIR	NR	Isobe et al. (2014)
Japan, Sea of Japan	3.74	Sea surface (75 cm)	0.1 – >10 mm	NR	NR	NR	Isobe et al. (2015)
China, southern estuaries	100–4,100	Sea surface (30 cm)	0.5–5 mm	PE, PP, PTFE, PVC	Raman microscopy	+	Zhao et al. (2015)
South Korea, Incheon/Kyeonggi coastal region	152,688	Surface microlayer (<400 µm)	50–2,000 µm	Paint particles, EPS, PET, PP, PVA, PVC, PVS	FTIR and FTIR microscopy	+	Chae et al. (2015)
	1,602	Sea surface (30 cm)	50–2,000 µm	Paint particles, EPS, PE, PET, PP, PVA, PVC, PVS			
South Korea, Jinhae Bay	12,600	Surface microlayer (150–400 µm)	<50–2,000 µm	Acrylic, paint resin particles, PE, PET, phenoxy resin, PL, PP, PS, PVC, synthetic rubber	FTIR	+	Song et al. (2015)
Hong Kong, coastal regions	0.51–279.9	Surface microlayer (153 µm)	0.03–4.96 mm	HDPE, LDPE, PP, PP – ethylene propylene, SA	ATR-FTIR	NR	Tsang et al. (2016)
Europe							
Portugal, coastal waters	0.002–0.036	Sea surface (20 cm)	>180 – > 325 µm*	Alkyd resin, polyacrylate, PE, PP	FTIR microscopy	NR	Frias et al. (2014)
French, Belgian and Dutch North Sea Coast	400	Sea surface (NR*)	30–300 µm	PS	Raman microscopy	+	Van Cauwenberghe et al. (2015)
Germany, Rostock coast (Warnemünde)	3,300	Sea surface (2–4 cm)	55 µm – 1 mm	NR	NR	+	Stolte et al. (2015)
North America							
Canada, Northern Pacific Ocean and Coastal British Columbia	8–9,200	Sea subsurface (4.5 m)	64.8–5,810 µm	NR	NR	NR	Desforges et al. (2014)
South America							
Brazil, Jurujuba Cove	16.4	Sea surface (20 cm)	< 1 mm – ≥5 mm	PE, PP	ATR-FTIR	NR	Castro et al. (2016)
Africa							
South Africa, south eastern coastline	257.9–1,215	Sea surface (45 cm)	0.080–5 mm	NR	NR	+	Nel and Froneman (2015)
Middle East							
Israel, Mediterranean Coast	7.68	Sea surface (10 cm)	<0.3–5 mm	NR	NR	NR	van der Hal et al. (2017)
Australia							
Coastal waters around Australia	0.007–0.053	Sea surface (0.17 m and 0.3 m)	0.4–82.6 mm	EVA, PE, PP, PS	ATR-FTIR	NR	Reisser et al. (2013)

Surface microlayer: the top of 1000 µm of the sea surface (Cunliffe et al. 2013). Sea subsurface: below the surface of the oceans, which is divided into five categories, i.e. epipelagic (from the surface to 200 m), mesopelagic (200–1,000 m), bathypelagic (1,000 m – 4,000 m), abyssopelagic (4,000 m – 6,000 m), and hadalpelagic (>6,000 m) (Akbari et al. 2017). EVA: ethylene vinyl acetate, PC: polycarbonate, PE: polyethylene, HDPE: high density polyethylene, LDPE: low density polyethylene, PET: polyethylene terephthalate, PL: polyester, PP: polypropylene, PS: polystyrene, EPS: expanded polystyrene, PTFE: polytetrafluoroethylene, PVA: polyvinyl alcohol, PVC: polyvinyl chloride, PVS: polyvinyl styrene, SA: styrene acrylonitrile, ATR-FTIR = Attenuated Total Reflection Fourier transform infrared, NR = Not Reported, * the size of microplastics in this study was based on the mesh size of nets used for collecting the water samples, QA = Quality Assurance, + = the study implemented measures to prevent microplastics contamination such as the analysis of blank samples, cleaning of all equipment and contact materials, and wearing 100% cotton coat during analysis.

Table 3. Summary of available data on microplastics detected in coastal fish species.

Location	Species	Habitat	Tissue	N and # individual ingested MPs (%)	Concentration (#/animal)[1]	Shape(s)	Type of polymer(s)	Identification method	QA	Ref.
Asia										
Indonesia, Poatere Fish Market Makassar	*Rastrelliger kanagurta* (Indian mackerel)	Pelagic	GIT	9 (55.5)	1.0 ± 1.1	fragments, film, monofilm	NR	Microscopic dissection	+	Rochman et al. (2015)
	Spratelloides gracilis (Blue sprat)	Pelagic	GIT	10 (40)	1.1 + 1.7	fragments				
	Siganus canaliculatus (White-spotted spinefoot)	Pelagic	GIT	3 (33.3)	0.3 ± 0.6	monofilaments				
Malaysia, local markets	*Chelon subviridis* (Greenback mullet)	Demersal	Viscera & gills	30 (30)	2	fragments, films, and filaments	PP, PE, PS, PET, nylon 6	Stereomicroscopy, Raman spectroscopy	+	Karami et al. (2017)
			flesh*		14					
	Johnius belangerii (Belanger's croaker)	Demersal	Viscera & gills	30 (40)	3					
			flesh*		13					
	Ratrelliger kanagurta (Indian mackerel)	Pelagic-neritic	Viscera & gills	30 (10)	2					
			Flesh*		1					
	Stolephorus waitei (spotty-face anchovy)	Pelagic-neritic	Flesh*	30 (5)	1					
China, Yangtze estuary	*Hyporhamphus intermedius* (Garfish)	Pelagic	GIT (stomach, gut)	18 (100)	3.7 ± 2.2	fibres, fragments and pellets	cellophane, PET, PL, etc.	Stereomicroscopy, FTIR-microscopy	+	Jabeen et al. (2017)
	Liza haemotocheila (Mullet)	Pelagic		18 (100)	3.3 ± 0.3					
	Coilia ectenes (Anchovy)	Pelagic		18 (100)	4.0 ± 1.8					
	Lateolabrax japonicas (Japan Sea Bass)	Pelagic		18 (100)	2.1 ± 0.3					
	Sillago sihama (Silver Sillago)	Pelagic		18 (100)	2.8 ± 1.5					
	Larimichthys crocea (Large yellow croaker)	Bentho-pelagic		18 (100)	4.6 ± 3.4					
	Psenopsis anomala (Pacific Rudderfish)	Bentho-pelagic		18 (100)	1.1 ± 0.3					
	Mugil cephalus (Black Mullet)	Demersal		18 (100)	3.7 ± 1.0					
	Terapon jarbua (Tiger Perch)	Demersal		18 (100)	3.0 ± 0.7					
	Sebastiscus marmoratus (False Kelpfish)	Demersal		18 (100)	4.2 ± 1.3					
	Photopectoralis bindus (Orangefin Ponyfish)	Demersal		18 (100)	4.1 ± 2.1					
	Thamnaconus septentrionalis (Greenfin filefish)	Demersal		18 (100)	7.2 ± 2.8					
	Oxyeleotrix marmorata (Marbled goby)	Demersal		18 (100)	4.2 ± 2.4					
Japan, Tokyo Bay	*Engraulis japonicas* (Japanese anchovy)	Benthic	GIT	64 (77)	2.3 ± 2.5	fragments, beads, filaments, foam	PP, PE, PS, E/P copolymer, E/P diene terpolymer	Microscopy, FTIR	+	Tanaka and Takada (2016)
India, Punnapara, Kerala	*Stolephorus commersonnii* (Commerson's anchovy)	Brackish, pelagic-neritic	Gut	16 (38)	NR	NR	NR	Microscopy	NR	Kripa et al. (2014)
Iran, Persian Gulf	*Alepes djedaba* (Shrimp Scad)	Pelagic, reef	Muscle	20 (NR)	0.8 ± 0.12[a]	fragment, fibre, pellet	NR	Microscopy, SEM	+	Akhbarizadeh et al. (2017a)

(*Continued*)

Table 3. (Continued).

Location	Species	Habitat	Tissue	N and # individual ingested MPs (%)	Concentration (#/animal)[1]	Shape(s)	Type of polymer(s)	Identification method	QA	Ref.
	Epinephelus coioides (Estuary Cod)	Benthic, brackish	Muscle	20 (NR)	0.78 ± 0.22[a]					
	Sphyraena jello (Pickhandle barracuda)	Pelagic, brackish	Muscle	15 (NR)	0.57 ± 0.17[a]					
	Platycephalus indicus (Bar-tailed Flathead)	Benthic	Muscle	16 (NR)	1.85 ± 0.5[a]					
Europe										
Greece, Eastern Aegean Sea	*Sphyraena viridensis* (Yellow Barracuda)	Epipelagic	Stomach	NR	42 ± 20.5	fibre	NR	Microscopy	NR	Miliou et al. (2016)
	Trachurus mediterraneus (Mediterranean Horse Mackerel)	Pelagic	Stomach		28 ± 19.5					
	Boops boops (Bogue)	Demersal	Stomach		15.4 ± 3.2					
Turkey, Mediterranean coast	*Argyrosomus regius* (Meagre)	Bentho-pelagic	GIT	51 (75)	2.47	fibres, hard plastics	Copolymer PS:isopropene, alloys (HIPPS/PP/PA6), PA resin, PE, PP	Stereomicroscopy, FTIR-microscopy	+	Guven et al. (2017)
	Caranx crysos (Blue Runner)	Reef	GIT	1 (100)	5					
	Dentex gibbosus (Pink Dentex)	Bentho-pelagic	GIT	14 (29)	1					
	Diplodus annularis (Annular Seabream)	Bentho-pelagic	GIT	48 (69)	2.85					
	Lithognathus mormyrus (Sand Seenbras)	Demersal	GIT	46 (35)	1.88					
	Liza aurata (Golden Grey Mullet)	Pelagic-neritic	GIT	39 (44)	7.47					
	Mullus barbatus (Blunt-Snouted Mullet)	Demersal	GIT	207 (66)	2.12					
	Mullus surmuletus (Striped Red Mullet)	Demersal	GIT	51 (65)	1.82					
	Nemipterus randalli (Randall's Threadfin Bream)	Demersal	GIT	135 (55)	2.24					
	Pagellus acarne (Axillary Seabream)	Bentho-pelagic	GIT	52 (67)	2.46					
	Pagellus erythrinus (Common Pandora)	Bentho-pelagic	GIT	54 (52)	1.21					
	Pagrus pagrus (Common Seabream)	Bentho-pelagic	GIT	9 (78)	1.86					
	Pelates quadrillineatus (Fourlined Terapon)	Reef	GIT	135 (65)	2.27					
	Pomadasys incises (Bastard Grunt)	Demersal	GIT	29 (55)	1.44					
	Sardina philcardus (European Philchard)	Pelagic-neritic	GIT	7 (57)	3.75					
	Saurida undosquamis (Brushtooth Lizardfish)	Reef	GIT	99 (55)	2.2					
	Sciaena umbra (Brown Meagre)	Demersal	GIT	1 (100)	3					
	Scomber japonicas (Pacific Chub Mackerel)	Pelagic-neritic	GIT	7 (71)	9.4					
	Serranus cabrilla (Comber)	Demersal	GIT	6 (67)	2.25					

(*Continued*)

Table 3. (Continued).

Location	Species	Habitat	Tissue	N and # individual ingested MPs (%)	Concentration (#/animal)[1]	Shape(s)	Type of polymer(s)	Identification method	QA	Ref.
	Siganus luridus (Dusky Spinefoot)	Reef	GIT	15 (87)	3.62					
	Sparus aurata (Gilthead Seabream)	Demersal	GIT	110 (44)	2					
	Trachurus mediterraneus (Mediterranean Horse Mackerel)	Pelagic-oceanic	GIT	98 (68)	2.58					
Spain, Atlantic and Mediterranean coast	*Mullus barbatus* (Blunt-Snouted Mullet)	Demersal	Stomach	128 (18.8)	1.9 ± 1.3	fibres, spheres, film, fragments	NR	Stereomicroscopy	+	Bellas et al. (2016)
Spain, Balearic Islands	*Boops boops* (Bogue)	Demersal	GIT	337 (57.8)	2.47–4.89	microfilaments	NR	Stereomicroscopy	+	Nadal et al. (2016)
Spain, ports of Mallorca Island (5 different ports)	*Mullus surmuletus Linnaeus 1758* (Striped Red Mullet)	Demersal	GIT	417 (27.3)		mostly filaments	PET, CA, polyacrylate, PAN, alkyd, polystyrene acrylonitrile methyl methacrylate	FTIR-microscopy	+	Alomar et al. (2017)
	(trawling net)		GIT		0.32 ± 0.04					
	(trammel net)		GIT		0.68 ± 0.10					
Spain, western Mediterranean Sea	*Boops boops* (Bogue)	Demersal	GIT	27	1.46 ± 0.66	mostly fibres	NR	Microscopic dissection	+	Ferrer et al. (2016)
	Sardina pilchardus (European Philchard)	Pelagic, near shore	GIT	17	1.43 ± 0.79					
	Engraulis encrasicolus (Anchovy)	Pelagic	GIT	17	1.18 ± 0.40					
				Total: 183 (17)						
Portugal, Atlantic coast	*Alosa fallax* (Twaite Shad)	Pelagic-neritic	GIT	1 (100)	1.0	particles and fibres	Particles: PP, PE, and alkyd resin. Fibres: rayon, PL, acrylic, nylon 6, and PE	Stereomicroscopy, FTIR-microscopy	+	Neves et al. (2015)
	Argyrosomus regius (Meagre)	Bentho-pelagic	GIT	5 (60)	0.8 ± 0.8					
	Boops boops (Bogue)	Demersal	GIT	32 (9)	0.09 ± 0.3					
	Dentex macrophthalmus (Large-eye Dentex)	Bentho-pelagic	GIT	1 (100)	1					
	Lophius piscatorius (Angler)	Bathy-demersal	GIT	2 (50)	0.5					
	Mullus surmuletus (Striped Red Mullet)	Demersal	GIT	1 (100)	2					
	Mullus surmuletus (Striped Red Mullet)	Demersal	GIT	3 (100)	1.66 ± 0.57					
	Scomber japonicas (Chub Mackerel)	Pelagic-neritic	GIT	35 (31)	0.57 ± 1.04					
	Scomber scrombus (Atlantic Mackerel)	Pelagic-neritic	GIT	13 (31)	0.46 ± 0.78					
France, Mediterranean Sea	*Engraulis encrasicolus, L.* (Anchovy)	Pelagic	Liver	13 (80)	NR	NR	PE, copolymer of styrene and acrylonitrile	Stereomicroscopy, Raman spectroscopy	+	Collard et al. (2017)
	Sardina pilchardus (European Philchard	Pelagic, near shore	Liver	2 (75)	NR					
	Clupea harengus (Atlantic Herring)	Pelagic, coastal	Liver	2 (75)	NR					
German, North Sea and Baltic Sea	*Limanda limanda* (Common Dab)	Demersal	GIT	89 (4.5)	0.03 ± 0.18	fibre, fragment	PE, PA, PP, PS, PET, PEST, PU	Stereomicroscopy, ATR-FTIR and FTIR-microscopy	+	Rummel et al. (2016)

(*Continued*)

Table 3. (Continued).

Location	Species	Habitat	Tissue	N and # individual ingested MPs (%)	Concentration (#/animal)[1]	Shape(s)	Type of polymer(s)	Identification method	QA	Ref.
	Platichthys flesus (Baltic Flounder)	Demersal	GIT	36 (5.6)						
	Gardus morhua (Atlantic Cod)	Brackish, bentho-pelagic	GIT	81 (1.2)	0.19 ± 0.61					
	Scomber scrombus (Atlantic Mackerel)	Pelagic-neritic	GIT	51 (17.7)						
Netherlands, Southern Bight, North Sea	*Sprattus sprattus* (Sprat)	Pelagic	GIT	400 (0.25)	2	spherical particles	PMMA/acrylic	ATR-FTIR	+	Hermsen et al. (2017)
Denmark, North Sea and Baltic Sea	*Gadus morhua* (Atlantic Cod, North Sea)	Brackish, bentho-pelagic	Stomach	28 (14)	NR	fragment, fibre	PU, PS, PP, PE	Raman spectroscopy	NR	Lenz et al. (2016a)
	Gadus morhua (Atlantic Cod, Baltic Sea)		Stomach	51 (16)						
	Clupea harengus (Atlantic Herring, North Sea)	Pelagic	Stomach	50 (30)						
	Clupea harengus (Atlantic Herring, Baltic Sea)		Stomach	55 (7)						
Sweden, coast	*Clupea harengus* (Atlantic Herring)	Pelagic	Stomach	130 (50)	0–53	fibres, fragments	NR	Stereomicroscopy	+	Ogonowski et al. (2017)
UK, Thames Estuary	*Platichthys flesus* (Baltic Flounder)	Demersal	GIT			fibres	PA, acrylic, nylon, PE, PET	Microscopic dissection, ATR-FTIR	+	Mcgoran et al. (2017)
	– Riverine 1			40 (90)	0.43 ± 0.75					
	– Estuarine			14 (71)	0.33 ± 0.49					
	– Riverine 2			12 (83)	0.85 ± 1.17					
	Osmerus eperlanus (Smelt)	Pelagic	GIT	10 (20)	0.2 ± 0.42					
Africa										
South Africa, KwaZulu-Natal, Durban Harbor	*Mugil cephalus* (Black Mullet)	Demersal	GIT	72.8	3.8 ± 4.7	fibres, fragments, films, mono-filaments, and twine	PS, others not clarified	Microscopic dissection	+	Naidoo et al. (2016)
North America										
USA, California, Half Moon Bay	*Atherinopsis californiensis* (Jack Silverside)	Pelagic	GIT	7 (28.6)	1.6 ± 3.7	fibres, fragment	NR	Microscopic dissection	+	Rochman et al. (2015)
	Engraulis mordax (Anchovy)				0.3 ± 0.5	fibres, film, monofilament				
		Pelagic	GIT	10 (30)						
	Morone saxatilis (Stripped Bass)	Pelagic	GIT	7 (28.6)	0.9 ± 1.2	film, fibre, foam				
	Oncorhynchus tshawytscha (Chinook Salmon)	Pelagic	GIT	4 (25)	0.25 ± 0.5	fibre				
	Ophiodon elongates (Lingcod)	Pelagic	GIT	11 (9.1)	0.1 ± 0.3	film				
Canada, eastern coast Newfoundland	*Gadus morhua* (Atlantic Cod)	Brackish, bentho-pelagic	GIT	205 (0.98)	1–2	fragment	NR	Visual and microscopic dissection	+	Liboiron et al. (2016)
South America										
Argentina, Río de la Plata estuary	*Luciopimelodus pati* (Pati)	Demersal	GIT	9 (100)	18.5 ± 18.9 (fibres) 0.7 ± 1.7 (others)	fibres, others (regular and irregular shapes)	NR	Stereomicroscopy	+	Pazos et al. (2017)

(*Continued*)

Table 3. (Continued).

Location	Species	Habitat	Tissue	N and # individual ingested MPs (%)	Concentration (#/animal)[1]	Shape(s)	Type of polymer(s)	Identification method	QA	Ref.
	Pseudoplatystoma corruscans (Spotted Sorubim)	Demersal	GIT	2 (100)						
	Oligosarcus oligolepis (Bocudo)	Pelagic	GIT	5 (100)						
	Parapimelodus valenciennis (Long-whiskered catfish)	Demersal	GIT	21 (100)						
	Odontesthes bonariensis (Pejerrey)	Brackish	GIT	1 (100)						
	Astyanax rutilus	Bentho-pelagic	GIT	12 (100)						
	Cyprinus carpio Common Carp)	Pelagic	GIT	2 (100)						
	Pimelodus maculatus (Long-whiskered catfis)	Bentho-pelagic	GIT	14 (100)						
	Prochilodus lineatus (Streaked Prochilod)	Bentho-pelagic	GIT	5 (100)						
	Hypostomus commersoni (Red Fin Uruguay)	Demersal	GIT	2 (100)						
	Cyphocharax voga (Characin)	Bentho-pelagic	GIT	14 (100)						
Brazil, Paraiba and Mamanguape estuaries	*Opisthonema oglinum* (Atlantic Thread Herring)	Pelagic	GIT	196 (9)	0.33	fibres, films, and fragments	NR	Stereomicroscopy	+	Vendel et al. (2017)
	Rhinosardinia bahiensis	Pelagic	GIT		0.35					
	Anchoa januaria (Rio Anchovy)	Pelagic-neritic	GIT		0.13					
	Lycengraulis grossidens (Atlantic Sabretooth Anchovy)	Pelagic-neritic	GIT		0.17					
	Atherinella brasiliensis (Brazilian Silverside)	Bentho-pelagic	GIT		0.03					
	Poecilia vivipara (Gargaru)	Bentho-pelagic	GIT		0.11					
	Hyporhamphus unifasciatus (Common Halfbeak)	Reefs	GIT		0.15					
	Oligoplites saurus (Leatherjacket)	Reefs	GIT		0.17					
	Diapterus auratus (Irish Mojarra)	Demersal	GIT		0.97					
	Diapterus rhombeus (Caitipa Mojarra)	Demersal	GIT		0.06					
	Eucinostomus argenteus (Silver Mojarra)	Reefs	GIT		0.02					
	Eugerres brasilianus (Brazilian Mojarra)	Demersal		GIT	0.06					
	Achirus lineatus	Reefs	GIT		0.5					
	Symphurus tessellatus (Tesselated Tonguefish)	Demersal	GIT		0.25					
	Sphoeroides testudineus (Checkered Puffer)	Reefs	GIT		0.09					
Brazil, Goiana Estuary	*Cynoscion acoupa* (Acoupa Weakfish)	Coastal, Demersal, Brackish	Stomach		0.08	filament	NR	Stereomicroscopy	+	Ferreira et al. (2016a)

(*Continued*)

Table 3. (Continued).

Location	Species	Habitat	Tissue	N and # individual ingested MPs (%)	Concentration (#/animal)[1]	Shape(s)	Type of polymer(s)	Identification method	QA	Ref.
	– juvenile: 14–220 mm)			469 (64)	1.8–2.5					
	– sub-adult: 220–340 mm)			25 (50)	2–8					
	– adult: >340 mm)			33 (100)	5.4–25.9					
Brazil, Goiana Estuary	*Stellifer brasiliensis* (Stardrum)	Demersal	Stomach	330 (6.9)	0.33 ± 0.08	fragments	PA	Stereomicroscopy	NR	Dantas et al. (2012)
	Stellifer stellifer (Little Croaker)	Brackish, Demersal	Stomach	239 (9.2)	0.52 ± 0.13					
Chile, Easter Island, Rapa Nui	*Decapterus muroadsi* (Amberstripe Cad)	Pelagic	GIT	20 (80)	2.5 ± 0.4	hard fragments, soft fragments, thread, film	PE, plastic paint (alkyd resins & epoxy-polyesters), PP	Microscopic dissection, ATR-FTIR and FTIR-microscopy	NR	Ory et al. (2017)
Mexico, estuary of the Gulf of Mexico	*Lutjanus campechanus* (Red Snapper)	Shallow waters, Reefs	GIT	2	NR	filament, fragment, film	NR	Microscopic dissection, FTIR	NR	Phillips and Bonner (2015)
	Lutjanus griseus (Mangrove Snapper)	Near shore, hard-bottom and coral	GIT	5						
	Lagodon rhomboides (Bream)	Vegetated bottom, mangrove	GIT	48						
	Sciaenops ocellatus (Red Drum)	Brackish, demersal	GIT	28						
	Cynoscion nebulosus (Spotted Seatrout)	Brackish, demersal	GIT	20						
	Paralichthys lethostigma (Southern Flounder)	Estuarine, coastal waters	GIT	8						
	Coryphaena hippurus (Common dolphinfish)	Open waters, near coastal areas	GIT	2						
				Total: 116 (10.4)						

PA: polyamide, PAN: polyacrylonitrile, PE: polyethylene, PET: polyethylene terephthalate, PL: polyester, PMMA: polymethylmethacrulate, PP: polypropylene, PS: polystyrene, PU: polyurethane; [1]: average, average ± standard deviation or range, N = the number of collected samples, NR = Not Reported, unkn = unknown, QA = Quality Assurance, + = the study implemented measures to prevent microplastics contamination such as the analysis of blank samples, cleaning of all equipment and contact materials, and wearing 100% cotton coat during analysis, *: flesh refers to all part of fish, including the bones, but do not include gills and viscera.

sampling, isolation and identification of microplastics may limit the comparability of the results.

Several studies implemented preventive measures to avoid sample contamination during analysis (Tables 1–4). This precautionary step is important since background contamination can occur due to plastics fibres from synthetic cloth, the usage of plastic equipment or insufficiently cleaned equipment, and poorly sealed samples. Procedures applied in preventing contamination include the application of procedural blanks, rinsed equipment, filtered solutions, clean laminar flow cabinet, coverage of samples, nitrile gloves, and the use of a cotton laboratory coat (Wesch et al. 2016). The implementation of strict quality assurance measures during microplastic analysis has been proved to be effective in preventing plastic contamination (Hermsen et al. 2017).

Sediment

Table 1 shows that sediment at the coastline of the Beibu Gulf (South China Sea) contains the highest number of microplastics, i.e. 5,020–8,720 particles/kg dw. An extremely high coastal population density and which produced the highest mass of mismanaged plastic waste in 2010, i.e. 1.32–3.53 MMT (Jambeck et al. 2015) are probably the most important factors explaining the microplastic pollution. The second and third most polluted coastal areas are Halifax Harbor – an urban estuary on the Atlantic coast of Canada, and the bay and beaches of Huatulco, Mexico. Both areas are also located close to human activities (Mathalon and Hill 2014; Retama et al. 2016).

The timing of the measurements can also play a role in determining the amount of microplastics in sediments as is demonstrated in the study on the Huatulco beaches in Mexico (Retama et al. 2016). The amount of microplastics fluctuated with tourism activity, resulting in higher levels of microplastics in sediment samples taken in December (tourism season) than in those taken in April. Hotels and sewage disposals in the proximity of beaches have been previously identified as potential sources of plastic debris (Cole et al. 2011).

Water column

Table 2 provides an overview of microplastics detected in the water column of coastal areas. The concentration ranges reported for the water column show more variation than in sediments, which is in line with the more dynamic character of the water column. Reported concentrations were highest in Incheon/Kyeonggi Bay and Jinhae Bay in South Korea. These areas are reported to be affected by microplastics originating from both aquatic (i.e., aquaculture, fishing activities, and international harbour) and terrestrial sources. The density of microplastics varied with water depth, i.e. the concentration in the surface microlayer was significantly higher than in water 30 cm below the surface (Chae et al. 2015).

Consistent with the pollution of the sediments, the level of microplastics in urban estuaries in China is relatively high (100–4,100 particles/m^3) (Zhao et al. 2015). This is in line with the high numbers of microplastics detected in sediment samples from Chinese coastal regions (Table 1).

Plastic types

The dominant types of polymers detected in sediment of coastal areas (Table 1) are polyethylene (PE), polypropylene (PP), polystyrene (PS), polyethylene terephthalate (PET) and polyamide (PA). The same polymer types are dominant in the water column (Table 2), together with polyvinylchloride (PVC). PE, PP and PA are widely used in textiles (Desforges et al. 2014) and fishing gear (e.g., ropes, nets, and fishing lines) (Claessens et al. 2011). PS, PVC and PET are primarily used as packaging materials, e.g., plastic bags, bottles, caps, films, containers, etc. (Claessens et al. 2011), but PE and PP are also used for this purpose. Of these five classes of commonly used polymers, PE, PP as well as the expanded form of polystyrene (EPS), have a lower density than sea water (1.02 g cm^{-3}), while PVC and PET, have a higher density. Taking only polymer density into account, one would expect PE, PP and EPS to dominate in the water column (Andrady 2015), and PVC and PET in the sediment (Galgani et al. 2015). However, this pattern does not emerge from the data presented in Tables 1 and 2 as low density polymers (i.e. PE, PP and EPS) are frequently

detected in sediments and conversely PVC and PET are found in the water column. This points towards the impact of other processes, i.e. floating plastics can be washed ashore and sedimented plastics can resuspend by strong currents (Chubarenko and Stepanova 2017). Furthermore, the density of microplastics may change due to biofouling and leaching of additives (Galgani et al. 2015).

Microplastic forms in coastal seafood

Tables 3 and 4 summarize the available data on microplastics detected in respectively coastal fish & shellfish species. From the data it is clear that a large variety of microplastic forms are present in a wide range of coastal seafood species. Dominant forms are fibres, fragments, films and filament, whereas spheres, pellets, foam, beads, twines, threads and flakes are less common. Unfortunately, since polymer identification is not frequently performed, only limited information is available on the polymer types found in fish and shellfish.

Commercially important fish

Microplastics are found in the gastrointestinal tract of both pelagic and demersal fish species that are commercially important for human consumption. The ingestion of microplastics by fish is mostly documented for predatory species, though some of the investigated species are primary filter feeding fish (Table 3). The amount of microplastics found in coastal fish species varies considerably between and within studies. The species-specific average values cover the range between 0 and 7.2 particles/animal. At least a part of this variation is caused by the fact that different tissues have been analysed. Some studies analysed the contents of the whole gut/gastrointestinal tract, while other studies merely analysed the contents of the stomach. Jabeen et al. (2017) compared the abundance of microplastics between stomach and gut from fish caught along the Chinese coast. In some of the 24 fish species analysed in this study, the abundance of plastics in the guts was higher than in the stomach. The authors suggest that the entire digestive tract should be analysed to obtain a realistic indication of the number of plastics to which animals are actually exposed (Jabeen et al. 2017).

Fish collected from the coastal areas of China are reported to contain the highest number of microplastics, i.e. 1.1–7.2 particles/animal. This finding is consistent with the high levels of microplastics found in coastal sediments and water of China (Tables 1 and 2) (Qiu et al. 2015; Zhao et al. 2015; Yu et al. 2016). Relatively high levels of microplastics were also detected in bogue (*Boops boops)* from the Balearic islands (2.47–4.89 particle/species) (Nadal et al. 2016) and the flathead grey mullet (*Mugil cephalus*) from Durban Harbor, South Africa (3.8 ± 4.7 particle/species) (Naidoo et al. 2016). Although data on microplastics in sediments and seawater of the Balearic Islands are lacking, the islands are subject to high levels of human activity such as commercial and recreational boating and coastal tourism (Naidoo et al. 2016). This may explain why the plastic pollution in that area is relatively high.

Several studies have pointed out that demersal fish species tend to contain more microplastics than pelagic species (Jabeen et al. 2017; Vendel et al. 2017). The data collected in this review (Table 3) confirm this as demersal species such as greenfin filefish (*Thamnaconus septentrionalis*), marbled goby (*Oxyeleotrix marmorata*), false kelpfish (*Sebastiscus marmoratus*), orangefin ponyfish (*Photopectoralis bindus*), black mullet (*Mugil cephalus*), tiger perch (*Terapon jarbua*), bogue (*Boops boops*), and Irish mojarra (*Diapterus auratus*) tend to have high microplastics levels. Demersal fish species live close to the seafloor where plastic litter accumulates. When feeding on benthic prey, some sediments will be swallowed together with the prey which increases the risk of ingesting plastic accidentally (Bellas et al. 2016).

Commercial shellfish and other seafood

Microplastics are widely found in shellfish species, such as bivalves and crustaceans (Table 4). Most studies focused on the blue mussel (*Mytilus edulis*) and oyster (*Crassostrea gigas*). Some studies also investigated microplastics in clam (*Venerupis philippinarum*), rock oyster (*Saccostrea forskalii*), and sea snail (*Littorina littorea*). Commercially important crustaceans such as brown shrimp (*Crangon crangon*), Norwegian lobster (*Nephrops norvegicus*), and crabs (*Carcinus maenas* and *Eriocheir*

sinensis) have also been found to ingest microplastics. Moreover, the presence of microplastics was also observed in echinoderms such as sea cucumber (Holothurian).

The abundance of microplastics is generally higher in shellfish than in fish. This can be attributed to the feeding strategy of shellfish, since most of them are filter feeders. Filter feeders, such as bivalves, oysters, and clams, display a non-selective feeding behaviour and are, therefore, more likely to ingest microplastics (Wesch et al. 2016). However, not all filter feeders are non-selective; the blue mussel (*Mytilus edulis*), for instance, displays selective feeding (i.e. selective feeding on particles of certain size) and particle rejecting behaviour (i.e. expelling particles as pseudofaeces) (Browne et al. 2008; De Witte et al. 2014; Van Cauwenberghe et al. 2015). Nevertheless, the highest numbers of microplastics were found in mussels from Halifax Harbor (Nova Scotia, Canada), i.e. 34–75 particles/animal. Considering Halifax Harbor Canada is one of the most polluted coastal areas with microplastics (Table 1), it is not surprising that mussels from that area contain high numbers of microplastics. This pattern can also be observed for China. The ranges of microplastics in shellfish varied between 4.3–57.2 particles/animal (2.1–10.5 particles/g ww) for 9 commercial bivalves collected from fishery farms or in situ along the coastal areas of China (Li et al. 2015), and 1.5–7.6 particles/animal (0.9–4.6 particles/g ww) in *M. edulis* from coastal areas of China (Li et al. 2016). Li et al. (2016) compared the abundance of microplastics in mussels from heavily contaminated areas and slightly contaminated areas along the coast of China, and concluded that mussels from heavily contaminated areas contain higher numbers of microplastics than those from less contaminated areas (Li et al. 2016).

Several studies have compared microplastics abundance in wild and cultured mussels, oysters, and clams. Those studies revealed that cultured shellfish tend to contain more microplastics than those sampled in situ. There are several possible explanations for this difference. In Canada, the use of plastic polypropylene lines in farm areas is considered the main reason for the higher number of microplastics in farmed mussels (Mathalon and Hill 2014). Another study linked the higher levels of microplastics in cultured *M. edulis* to fishery activities in the harbors and ports, such as fishing net repair and dumping of old nets (De Witte et al. 2014). A study in British Columbia, Canada, found that manila clams (*Venerupis philippinarum*) from shellfish farms had more plastic fibres than manila clams collected from beaches, but this difference was not significant (Davidson and Dudas 2016). Contrasting results were obtained by a study in China, which revealed that farmed mussels were less polluted (1.6 particles/g) than wild mussels (2.7 particles/g). The farmed mussels in China were cultured in areas with good water quality (i.e., areas less affected by human activities), while the wild mussels were taken from highly polluted coastal areas (Li et al. 2016). These results indicate that the level of microplastics pollution in the marine environment is an important factor determining the abundance of microplastics in mussels.

In addition to selective feeding and rejection behaviour (of *M. edulis*), bivalves also have the ability, when transferred to clean sea water, to depurate contaminants from their guts, including microplastics (GESAMP 2016). So far, only one study has examined the potential reduction of microplastics in bivalves through depuration (Van Cauwenberghe and Janssen 2014). The study showed that after three days of depuration the levels of microplastics in blue mussel (*M. edulis*) taken from the North Sea (Germany) reduced from 0.36 ± 0.07 particles/g ww to 0.24 ± 0.07 particles/g ww. The same trend was observed in a study on pacific oyster (*C. gigas*) from Brittany, France. This study determined a decrease of microplastics levels after a three days-depuration period from 0.47 ± 0.16 particles/g ww to 0.35 ± 0.05 particles/g ww. It can be concluded that, although depuration reduces the number of microplastics in shellfish, most particles seem to remain in the animals. Further research is needed to determine the impact of depuration on the removal of microplastics more precisely, particularly to determine the time required by bivalves to remove all or most of the microplastics.

Trophic transfer

Microplastics can accumulate within the food chain. Although only limited data is available, experimental evidence, mainly from laboratory-controlled studies, confirms that microplastics can be transferred

Table 4. Summary of available data on microplastics detected in coastal shellfish species.

Location	Species	Habitat	Tissue	N and # individual ingested MPs (%)	Concentration (#/animal)[1]	Concentration (#/gram ww)[2]	Shape(s)/ Form (s)	Type of polymer(s)	Identification method	QA	Ref.
Asia											
China, Shanghai	9 commercial bivalves	Fishery farms Coastal waters	Soft tissue	NR	4.3–57.2	2.1–10.5	fibres, fragments, pellets	NR	FTIR-microscopy	+	Li et al. (2015)
China, coastline	*Mytilus edulis* (Blue mussel)	Intertidal shallow water – Farm – Wild	Soft tissue	NR	1.5–7.6 NR NR	0.9–4.6 1.6 2.7	fibres, fragments, spheres, flakes	CP, PET, PL	FTIR-microscopy and SEM	+	Li et al. (2016)
Thailand, upper Gulf of Thailand	*Saccostrea forskalii* (Rock Oyster)	Intertidal, rocky shores	Soft tissue	NR	NR	0.37–0.57	rod shape and fragmented fibres	PA, PET, PS	Raman spectroscopy	NR	Gamage et al. (2017)
	Littoraria sp (Periwinkle)	Mangrove	Soft tissue	NR	NR	0.17–0.23					
Europe											
Belgium, coastline	*Mytilus edulis* (Blue mussel)	Intertidal shallow water – Farm – Wild	Soft tissue	NR	 NR NR	 0.35 0.26–0.51	fibres	NR	Stereomicroscopy	+	De Witte et al. (2014)
France, Atlantic coast	*Mytilus edulis* (Blue mussel)	Intertidal shallow water	Soft tissue	NR	0.61 ± 0.56	0.23 ± 0.20	fragments, filaments	PP, PE, PE-PP copolymer, PS, PMMA, PL, ABS	FTIR-microscopy	+	Phuong et al. (2017)
	Crassostrea gigas (Pacific Oyster)	Estuaries	Soft tissue	NR	2.10 ± 1.71	0.18 ± 0.16	fragments, filaments	PE, PP, ABS, PE-PP copolymer, PL, PIB, PS			
Germany, North Sea	*Mytilus edulis* (Blue mussel)	Intertidal shallow water – pre depuration – post depuration	Soft tissue	NR	NR NR	0.36 ± 0.07 0.24 ± 0.07	spheres	NR	µ -Raman spectroscopy	+	Van Cauwenberghe and Janssen (2014)
France, Brittany	*Crassostrea gigas* (Pacific oyster)	Estuaries – pre depuration – post depuration	Soft tissue	NR	NR NR	0.47 ± 0.16 0.35 ± 0.05	spheres	NR	µ -Raman spectroscopy	+	Van Cauwenberghe and Janssen (2014)
Netherlands, North Sea coast	*Carcinus maenas* (Littoral Crab)	Estuaries, intertidal zone, pelagic	Soft tissue	0%	NR	0	fibres, foils, and spheres	NR	FTIR-microscopy	+	Leslie et al. (2017)
	Mytilus edulis (Blue mussel)	Intertidal shallow water	Soft tissue	NR		13.1					
	Crassostrea gigas (Pacific Oyster)	Estuaries	Soft tissue	NR		2.4–10.9					
	Littorina littorea (Periwinkle)	Rocky coasts, mud flats, estuaries	Soft tissue	NR		2.5–3.8					
France, Belgium & Netherlands, North Sea coast	*Mytilus edulis* (Blue mussel)	Intertidal shallow water	Soft tissue	NR	NR	0.2 ± 0.3	spheres	LDPE, HDPE, PS	µ -Raman spectroscopy	+	Van Cauwenberghe et al. (2015)
Poland, Baltic Sea coast	*Eriocheir sinensis* (Chinese Mitten Crab)	Estuaries	Stomach	9–28%	NR	NR	balls, strands	NR	Microscopy	NR	Wójcik-Fudalewska et al. (2016)
Scotland, Clyde Sea	*Nephrops norvegicus* (Norway Lobster)	Muddy bottoms	GIT	83%	NR	NR	balls, strands, strands and balls	PP	Microscopy, SEM, Raman spectroscopy	+	Murray and Cowie (2011)
Europe, Southern North Sea & Channel	*Crangon crangon* (Brown Shrimp)	Benthic	Whole	63%	1.23 ± 0.99	0.68 ± 0.55	fibres	NR	Stereomicroscopy and hot needle test	+	Devriese et al. (2015)

(*Continued*)

Table 4. (Continued).

Location	Species	Habitat	Tissue	N and # individual ingested MPs (%)	Concentration (#/animal)[1]	Concentration (#/gram ww)[2]	Shape(s)/ Form (s)	Type of polymer(s)	Identification method	QA	Ref.
North America											
USA, Californian market	*Crassostrea gigas* (Pacific Oyster)	Estuaries	Soft tissue	33.3%	1.0 ± 1.1	NR	fibres	NR	Microscopic dissection	+	Rochman et al. (2015)
Canada, British Columbia, Baynes Sound	*Venerupis philippinarum* (Manila Clam)	Intertidal zone, estuaries/ brackish • Wild • Farm	Soft tissue	41% 59%	8.4 ± 8.5 11.3 ± 6.6	0.9 ± 0.9 1.7 ± 1.2	fibres, films, fragments	NR	Microscopy	+	Davidson and Dudas (2016)
Canada, Nova Scotia, Halifax Harbor	Mussels (unspecified)	• Wild • Farm	Soft tissue	NR NR	34 75	NR NR	filaments, fragments, film	PP, PET, polymethacry-late, PS, PA	Microscopic dissection	+	Mathalon and Hill (2014)
Equatorial mid-Atlantic	Holothurian (sea cucumber)	Benthic	Stomach, mouth, gills	NR	2	NR	microfibres	Modified acrylic, PP	Polarized light microscopy	+	Taylor et al. (2016)

CP: cellophane, PA: polyamide, PE: polyethylene, LDPE: low density polyethylene, HDPE: high density polyethylene, PET: polyethylene terephthalate, PIB: polyisobutylene; PL: polyester, PMMA: polymethylmethacrulate, PP: polypropylene, PS: polystyrene, PU: polyurethane; [1]: average, average ± standard deviation or range, N = the number of collected samples, NR = Not Reported, unkn = unknown, QA = Quality Assurance, + = the study implemented measures to prevent microplastics contamination such as the analysis of blank samples, cleaning of all equipment and contact materials, and wearing 100% cotton coat during analysis.

between trophic levels (Carbery et al. 2018). The trophic transfer of microplastics have been proven by a field study (Miliou et al. 2016) and laboratory experiments (Farrell and Nelson 2013; Tosetto et al. 2017). Setälä et al. (2014) reported the uptake of 10 µm fluorescent polystyrene microspheres in several Baltic Sea zooplankton taxa and demonstrated the potential of trophic transfer via other planktonic species. Trophic transfer has also been observed between beach hoppers, frill gobies and ray-finned fish (Tosetto et al. 2017), and between mussels (*M. edulis*) and crabs (*C. maenas*) (Farrell and Nelson 2013). A similar study using mussels (*Perna perna*) as prey and puffer fish (*Spheoeroides greeleyi*) and crab (*Callinectes ornatus*) as predators, revealed the presence of microplastic particles in the faeces of the predators, but only when fed contaminated mussels. Notwithstanding the confirmed transfer of microplastics to both fish and crabs, the risk to higher trophic levels can be considered as negligible due to the rapid depuration of the microplastics (Santana et al. 2017).

Concluding, there are sufficient indications that trophic level transfer may lead to accumulation in higher trophic levels (GESAMP 2016). This triggers concerns about human health since fish and shellfish of higher trophic levels are often on our menu. This holds particularly for the consumption of shellfish, such as bivalves, and small fish like anchovies, since these species are eaten whole (GESAMP 2016). However, the implications of trophic transfer of microplastics for higher predators and eventually humans are still poorly understood (Carbery et al. 2018).

Uptake and toxicity of microplastics in coastal seafood species

Table 5 provides an overview of toxicity assessments performed on coastal fish and shellfish. Fish species have been exposed to plastic polymers, such as PE, PVC and LDPE, in various sizes and concentrations, whereas PS, PP and HDPE have been used in shellfish studies. All types of plastics used in experimental toxicity studies are widely produced and used (Barnes et al. 2009).

Several intestinal uptake mechanisms for nano-sized and micro-sized particles up to 150 µm are described for vertebrates (Volkheimer 1977;

Powell et al. 2010; Van Cauwenberghe and Janssen 2014; Bouwmeester et al. 2015). In addition, laboratory studies with vertebrate species, including fish have demonstrated plastic particle translocation. For instance, translocation of PS nanoparticles (53 nm & 180 nm) has been observed in the brain of Crucian carp (*Carassius carassius*) (Mattsson et al. 2017) while Collard et al. (2017) demonstrated the translocation of microplastic particles (323 ± 101 µm) in the liver of commercial species of European anchovies (*Engraulis encrasicolus*), Atlantic herring (*Clupea harengus*) and European pilchard (*Sardina pilchardus*). However, further research is needed to assess the exact translocation pathways of nano- and micro-sized particles (Collard et al. 2017).

Based on the reported data on microplastics in wild seafood species, it has been argued that the likelihood of translocation in fish is small (Jovanović 2017). However, it should be kept in mind that translocation of microplastic particles measuring less than 150 µm may be underreported due to the lack of analytical methods capable of characterizing and quantifying small-sized plastic particles from samples taken from wild-caught animals (GESAMP 2016).

Several dietary toxicity studies with microplastics have been performed on fish species. Ferreira et al. (2016b) exposed common goby (*Pomatoschistus microps*) to PE particles (1–5 µm), alone or in combination with gold nanoparticles. The PE particles did not influence the uptake or the toxic effects of the gold nanoparticles, nor were any PE particle-related changes detected in the predatory performance of the fish. However, ethoxyresorufin-O-deethylase (EROD) and glutathione-S-transferase (GST) assays revealed a significantly increased phase I biotransformation activity and increased oxidative stress in the PE particle-exposed group under increased temperature (from 20 °C to 25 °C). A study with larvae of the European bass (*Dicentrarchus labrax*) revealed that the ingestion of PE microbeads (10–45 µm) for 7–43 days increased cytochrome P450 1A1 (*cyp1a1*) expression and caused obstruction of the gastrointestinal tract, although no significant effects on larval growth were observed. The highest exposure concentration (12 mg PE/g diet) led to increased mortality during the first stages of sea bass larval development, which was attributed to the narrow diameter of the esophageal, gastric and intestinal lumens of the fish larvae (Mazurais et al. 2015). The ingestion of PVC particles (<300 µm) at 0.1% w/w of feed by adults of European bass for 90 days resulted in structural and functional deterioration of the distal gut, but did not increase mortality (Peda et al. 2016). A study in which Japanese rice fish (*Oryzias latipes*) were fed PE pellets (<500 µm; 10% w/w of diet) found a reduced choriogenin (Chg H) gene expression in male fish, and a reduced vitellogenin (Vtg I), Chg H and estrogen receptor gene expression in female fish, which are early signs of endocrine disruption (Rochman et al. 2014). Exposure to 0.1 g L^{-1} of 53 nm PS nanoparticles caused morphological changes in the brain and affected hunting and feeding behaviour in Crucian carp (*Carassius carassius*) while exposure to 0.1 g L^{-1} of 180 nm PS particles revealed faster feeding and higher activity (Mattsson et al. 2017). Exposure of developing zebra fish (*Danio rerio)* to 51 nm PS nanoparticles (0.1, 1, or 10 mg kg^{-1}) from 6 hours to 120 h post-fertilization (hpf), revealed accumulation in the yolk at 24 hpf followed by migration to the gastrointestinal tract, gallbladder, liver, pancreas, heart, and brain during 48–120 hpf. The exposure did not cause significant mortality, morphological deformities or changes in mitochondrial metabolism but decreased heart rate in embryos and induced larval swimming hypoactivity. A depuration phase (120–168 hpf) decreased the concentration in all organs, although at a slower rate in the pancreas and the gastrointestinal tract (Pitt et al. 2017).

Several studies have focused on the impact of PS particles on shellfish, and in particular on the blue mussel (*Mytilus edulis*). As mentioned before, *M. edulis* is a selective filter feeder, ingesting only algae and particles of appropriate size and shape. Larger particles are selectively expelled as pseudofaeces (Defossez and Hawkins 1997; Ward and Shumway 2004). In congruence, an 8 hours exposure to 30 nm PS particles (at 0, 0.1, 0.2 and 0.3 mg mL^{-1}) resulted in reduced filtering activity and the production of nanopolystyrene containing pseudofaeces (Wegner et al. 2012). In addition, by simultaneously exposing *M. edulis* to 10, 30, and 90 µm PS particles (110 particles/mL for 14 days), Van Cauwenberghe et al. (2015) showed that the smallest particles are more easily retained than the larger

particles. Furthermore, Kholandhasamy et al. (2018) revealed that adherence of microfibers to the surface of soft tissues of *M. edulis* contributes strongly to the accumulation, particularly in organs not involved in ingestion processes. Translocation of particles from the gut to the circulatory system of *M. edulis* was demonstrated for PS microspheres with sizes of 3 and 9.6 µm (Browne et al. 2008).

A number of studies demonstrated that microplastics have the potential to induce a variety of physiological effects in bivalves. Exposure of *M. edulis* to HDPE particles (0–80 µm) at 2.5 mg mL^{-1} resulted in an inflammatory response of the digestive tract after 3 hours of exposure as indicated by granulocytoma formation at tissue level and decreased stability of lysosomal membrane at cellular and subcellular levels (von Moos et al. 2012). Exposure of the Mediterranean mussel (*Mytilus galloprovincialis*) to PE and PS particles <100 µm (both in the form of virgin particles and contaminated with pyrene) at 1.5 g L^{-1} for 7 days caused alterations of immunological responses, the lysosomal compartment, peroxisomal proliferation, the antioxidant system, neurotoxic effects, onset of genotoxicity and changes in gene expression profile (Avio et al. 2015). Exposure of *Mytilus spp*. for 7 days to PS microbeads (with size 2 µm and 6 µm, both virgin particles at 32 g L^{-1} and contaminated with fluoranthene particles at 30 g L^{-1}) and followed by depuration, resulted in increased haemocyte mortality and triggered substantial modulation of the cellular oxidative balance (i.e., increased reactive oxygen species production in haemocytes, and increased antioxidant and glutathione-related enzymes in mussel tissues) (Paul-Pont et al. 2016). A study using oyster species showed that the exposure of PS spheres (with size 2 µm and 6 µm at 0.023 mg L^{-1}) to *Crassostrea gigas* for 2 months showed increased food consumption, reproductive disruption (significant decreases in oocyte number, diameter and sperm velocity), and reduction of offspring performance during larval stages (Sussarellu et al. 2016). The higher food consumption by the oyster suggests an increase of stress and energy demand to maintain homeostasis (Sussarellu et al. 2016). *Atactodea striata*, a clam commonly found at the coast in Hong Kong, was fed 0, 10, 1000 items/L PS microgranules sized between 63 µm and 250 µm for 10 days. While the respiration rate and absorption efficiency remained unchanged, the highest concentration caused a reduction in the clearance rate which could reduce the energy intake. However, it was noted that the production of pseudofaeces and faeces, and depuration in clean water, effectively limited the ingestion and retention of microplastics, resulting in low amounts of microplastics in the body (Xu et al. 2017).

A few studies reported on uptake and toxicity of microplastics in crustaceans. In a dietary study, Brennecke et al. (2015) showed that particles larger than 150 µm can be translocated in the mudflat fiddler crab (*Uca rapax*). The ingested PS particles (180–250 µm at 108–1000 mg kg^{-1} of dry sediment) were subsequently detected in the gills, stomach and hepatopancreas. Dietary exposure of the Norway lobster (*Nephrops norvegicus*) to PCB-spiked PE and PS microspheres (6 µm, 500–600 µm) at 155 mg/9.64 g of gelatin cubes for three weeks followed by one week of depuration, had no impact on the nutritional state of wild *Nephrops*, but revealed a limited accumulation of PCBs in tail tissue for PE particles of 500–600 µm (Devriese et al. 2017). Dietary exposure of the shore crab *Carcinas maenas* to PP fibres (1–5 mm in length) for 4 weeks showed reduced food consumption from 0.33 to 0.03 g d^{-1} and significant reduction of available energy for growth from 0.59 to −0.31 kJ d^{-1} in crabs fed with 1% plastic (Watts et al. 2015), suggesting a depletion of energy storage.

Studying the presence and effects of nano- and microplastics in planktonic animal species that have a pivotal role in the food chain can provide insight into the detrimental effects of plastic pollution through the aquatic food web. Dietary exposure of the water flea (*Daphnia magna)*, a planktonic freshwater crustacean, to a range of polymeric nanoparticles revealed toxic effects for 52 nm-sized amino-modified positively charged PS nanoparticles (> 0.075 g L^{-1}, 13 hours exposure), whereas larger particles of the same material (120–330 nm, at 0.025–0.15 g L^{-1}) did not induce any observable effects. Interestingly, indirect intake via algal food was higher than direct intake from water (Mattsson et al. 2017). Larval stages of

brine shrimp (*Artemia franciscana*) and barnacle (*Amphibalanus Amphitrite*), two marine planktonic crustaceans, were exposed for 24 and 48 h to 0.1 µm PS beads at concentrations ranging from 0.001 to 10 mg L^{-1}. The results showed accumulation of PS beads in the gut of both crustaceans after 24 and 48 hours. It was noted that brine shrimps constantly ingested and excreted microbeads whereas barnacle did not excrete microplastics. No significantly increased mortality was observed (in both larvae), but exposure to high concentrations (>1 mg L^{-1}) for 48 hours changed swimming activity and increased activity of cholinesterase and catalase, which are indicative for neurotoxic and oxidative stress (Gambardella et al. 2017). In a study characterizing the size- and shape-dependent effects of microplastic particles, adult daggerblade grass shrimp (*Palaemonetes pugio*) was exposed for 3 h to 30, 35, 59, 75, 83, 116 and 165 µm PE or PS spheres and to 34 and 93 µm PP fragments and to 34 and 93 µm PP fibres at a concentration of 50,000 particles/L (Gray and Weinstein 2017). The results revealed that ingestion and ventilation are the main uptake pathways and that the number of ingested particles is influenced by the shape (i.e. fragments > spheres > fibres). The residence time of the particles in the digestive tracts and gills was 43.0 ± 13.8 hours and 36.9 ± 5.4 hours, respectively. Spheres and fragments smaller than 50 µm were not acutely toxic, while for sizes above 50 µm the mortality for the three shapes ranged from 5% to 40%, with a significantly higher mortality (55%) for 93 µm fibres.

Microplastics can act as vectors for adventitious chemical contaminants such as monomers (i.e. the building blocks of plastics), additives (including plasticizers, flame retardants, lubricants, UV-stabilizers, hydrocarbons, antioxidants, as well as intermediates and compounds for dyes and inks) and contaminants absorbed from the environment. The absorption of persistent organic pollutants (POPs) such as polychlorinated biphenyls (PCBs), polycyclic aromatic hydrocarbons (PAHs), petroleum hydrocarbons, dichlorodiphenyltrichloroethane (DDT), organochlorine pesticides (hexachlorocyclohexanes, hexachlorobenzene, chlordanes, and mirex), organophosphorus esters, and phthalates by plastics materials has been widely documented (Browne et al. 2013; Rochman et al. 2013; Avio et al. 2015; Batel et al. 2016; Jang et al. 2016; Wardrop et al. 2016; Kwon et al. 2017; Zhang et al. 2017). A recent study using *Mytilus spp*. found that 7 days of exposure to PS microbeads in combination with fluoranthene resulted in a higher fluoranthene concentration in mussels after 7 days of depuration compared to mussels exposed to fluoranthene alone (Paul-Pont et al. 2016). This phenomenon may be related to direct effects of PS microbeads on detoxification mechanisms, impairment of filtration activity, or to the remaining PS microbeads in the gut (Paul-Pont et al. 2016).

Recent findings indicate that chemicals sorbed to microplastics, such as brominated flame retardants (e.g., PBDEs (Wardrop et al. 2016) and HBCDs (Jang et al. 2016)), nonylphenol, triclosan, PAHs and PCBs, can be transferred to organisms and then trigger adverse effects (Browne et al. 2013; Rochman et al. 2013; Avio et al. 2015). Bioaccumulation of chemical pollutants in marine biota via food web transfer has also been reported (Batel et al. 2016). However, both field and modelling studies suggest that transfer of environmental pollutants through microplastics is negligible compared to other routes of uptake (Gouin et al. 2011; Bakir et al. 2016; Espinosa et al. 2016; Koelmans et al. 2016; Ziccardi et al. 2016; Lohmann 2017; Smith et al. 2018). Nonetheless, caution seems warranted since many of the chemicals sorbed to microplastics are known to be potent toxicants in humans and marine biota, triggering adverse effects such as endocrine disruption, neurological disorders, and reduced reproductive success (GESAMP 2016).

In the marine or coastal environment, microplastics may also interact with microorganisms. For instance, a microcosm experiment from Harrison et al. (2014) investigated the potential of microplastics in sediment to function as sites of attachment for naturally occurring bacteria. The results revealed that LDPE microplastics were colonized within 7 days and that after 14 days, these plastisphere communities were dominated by the genera *Acetobacter* and *Colwellia* (Harrison et al. 2014). The presence of potentially pathogenic hitchhikers such as *Vibrio spp*. was demonstrated on certain types of

microplastics (PE, PP, and PS fragments) and in water collected from North/Baltic Sea (Kesy et al. 2016; Kirstein et al. 2016). From all collected plastic particles, pathogenic bacteria *V. parahaemolyticus* were identified on 12 microplastic fragments. Although the occurrence of microorganism assemblages and hitchhikers has been proved by those studies, the consequences for the respective ecosystems remain unknown.

Intake of microplastics via human consumption of seafood

Since seafood constitutes an important food source, especially for coastal communities, there is an urgent need to assess the potential risks involved. Estimation of dietary exposure is an important first step. Table 6 presents the estimated intake of microplastics from seafood based on the amounts of seafood consumed globally in 2013 (FAO 2017a) and the concentrations of microplastics reported in Tables 3 and 4. Since the spatial variation in microplastics concentrations is high, even within the same continent, we used the minimum and maximum concentration reported in each continent. The estimated intake can reach up to 66 x 10^3, 28 × 10^3 and 36 × 10^3 particles/day through fish, crustacean, and molluscs consumption, respectively (Table 6). This is higher than the values estimated by Vandermeersch et al. (2015) and Van Cauwenberghe and Janssen (2014), which range from 365 up to 11 × 10^3 particles/year (Van Cauwenberghe and Janssen 2014).

Data in Table 6 represents a conservative scenario of dietary exposure to microplastics from fish and shellfish in general. Estimating microplastic intake via fish consumption (Table 6) is complex, because microplastic particles found in fish are mostly found in digestive tracts, which are usually discarded. Degutting can minimize the direct exposure to microplastics. However, small fish species such as anchovies, are eaten whole. Therefore, exposure assessment based on microplastics abundance found in the digestive tract of fish is still important, since data on microplastics found in the tissue of fish are still very limited. This approach is necessary for scientific purpose, particularly to portray the risk level of microplastics exposure to human at current condition.

Discussion

Methodological considerations

The growing research interest in microplastics has resulted in abundant occurrence data on microplastics in the environment and seafood (Tables 1–4). However, the lack of standardized methods and quality assurance protocols hinders interpretation of the results and comparisons between studies. This raises the question whether results are representative and comparable. The establishment of harmonized protocols covering sampling, pre-treatment, analytical techniques and quality assurance is required to obtain unbiased and comparable data on the presence of microplastics in the environment.

Quality assurance

The omnipresence of (micro)plastics in natural and human environments implies a serious contamination risk during sampling and analysis. For example, synthetic microfibres have been shown to be abundant in both indoor and outdoor air (Dris et al. 2016, 2017), resulting in contaminated samples during microplastics analysis (Wesch et al. 2017). Performing microplastic analysis under clean air conditions has been proven significantly to reduce the abundance of small fibres and give more reliable results (Foekema et al. 2013; Hermsen et al. 2017). Inclusion of blank and spiked samples during analysis is an effective way to assure the validity of the data obtained (Rummel et al. 2016; Hermsen et al. 2017).

Of the 72 papers included in the present review (Tables 1–4), approximately 60% implemented some kind of quality assurance, although the extent of the quality control measures varied substantially between studies. Only 55% of 72 studies ran blank samples, 28% checked background contamination, and 17% ran the combination of blank samples and spiking method or background checking. The development, implementation and harmonization of strict quality assurance criteria are absolutely essential to improve the accuracy of analytical data on microplastics.

Table 5. Laboratory experiments exposing coastal seafood species to microplastics and the observed effects.

	Species	Polymer type & shape	Size	Exposure route: concentration; duration	Endpoint(s)	Observed effect(s)	Reference
Fish	*Pomatoschistus microps*	PE spheres alone or in combination with gold nanoparticles.	1–5 µm	*Water*: 0.184 mg L^{-1}; 96 hours	Head and body of individual fish	Exposure to PE spheres did not change uptake and toxicity of gold nanoparticles, nor did it significantly change predatory performance, but it did induce a temperature depend increase of phase I biotransformation activity and oxidative stress.	Ferreira et al. (2016b)
	Dicentrarchus labrax larvae	fluorescent PE microbeads	10–45 µm	*Dietary*: 0, 0.12, and 1.2% (w/w) diet; 7–43 days	Larvae	Significant positive correlation between number of PE beads scored per larvae at 20 days after hatching (dph) and cytochrome P450-1A1 (*cyp1a1*) expression. Exposure to the highest concentration caused a slightly increased mortality, but in general, there was only limited impact of PE microbeads on sea bass larvae.	Mazurais et al. (2015)
	Dicentrarchus labrax	PVC (native and polluted)	<0.3 mm	*Dietary*: 0.1% (w/w) of feed; 90 days	Gut	Gradual histopathological alterations varying from moderate to severe related to exposure times, indicating structural and functional deterioration of in particular the distal part of the gut.	Peda et al. (2016)
	Oryzias latipes	PE (pellets and marine-plastic treatment)	<0.5 mm	*Dietary*: 10% (w/w) of diet; 1–2 months	Liver and gonads	Decreased choriogenin (Chg H) gene expression in male fish and decreased vitellogenin (Vtg I), Chg H and estrogen receptor gene expression in female fish suggests the capability of inducing endocrine disrupting effects.	Rochman et al. (2014)
	Carassius carassius	PS-NH2 amino modified particles	53 and 180 nm	*Water*: 0.1 g L^{-1} (with particles size 180 and 53 nm); 0.029 g L^{-1} (particle size 53 nm); 0 g L^{-1} (control). The fish were also fed *Daphnia magna* that had ingested PS particles from contaminated algae (60 individuals to each aquarium consisted of 3 fish). The fish were fed *Daphnia magna* every 3 days. Exposure: 67 days	Brain	The transfer of NPs through the food chain (from algae through zooplankton to fish) was demonstrated and the accumulation of PS particles caused morphological changes in brain and affected hunting and eating behaviour.	Mattsson et al. (2017)
	Danio rerio (embryo and larvae)	PS nanoparticles	51 nm	*Water*: 0, 0.1, 1, 10 mg L^{-1} 168 hours	embryo and larvae (GI tract, head, gall bladder, liver, pericardium, pancreas)	Migration of PS particles from yolk to other organs throughout embryo development. The exposure of nano-PS to the embryo did not cause significant mortality, morphological deformities and mitochondrial bioenergetics, but decreased the heart rate in embryos and induced larval swimming hypoactivity.	Pitt et al. (2017)
Shellfish	*Mytilus edulis*	PS spheres	30 nm	*Water*: 0, 0.1, 0.2, 0.3 mg mL^{-1}; 8 hours	Tissues, pseudofaeces	Increased production of pseudofaeces and reduced filtering activity.	Wegner et al. (2012)
	Mytilus edulis	PS spheres	10, 30, 90 µm	*Water*: 110 particles/mL; 14 days	Digestive tract	No significant effects on the overall energy budget.	Van Cauwenberghe et al. (2015)

(*Continued*)

Table 5. (Continued).

Species	Polymer type & shape	Size	Exposure route: concentration; duration	Endpoint(s)	Observed effect(s)	Reference
Mytilus edulis	PS spheres	3, 9.6 µm	*Water*: 0.51 mg mL^{-1}; 12 hours	Digestive tract, haemolymph	Translocation to circulatory system within 3 days, with a maximum accumulation at 12 days and persistence for over 48 days. No significant effects on the oxidative status of haemolymph, the viability and phagocytic activity of haemocytes, or filter-feeding activity.	Browne et al. (2008)
Mytilus edulis	HDPE grain	0–80 µm	*Water*: 2.5 mg mL^{-1}; 3–96 hours	Digestive tract, haemolymph	Uptake in the stomach, transfer into the digestive gland and accumulation in the lysosomal system; exposure causes inflammatory response in digestive tract demonstrated by granulocytoma formation and lysosomal destabilization	von Moos et al. (2012)
Mytilus edulis	Microfibres	0.05–5 mm	*Water*: 2,000 microfibers/L; 48 h exposure and 48 h elimination	Gills, gut, stomach, mantle, gonad, adductor, visceral tissues, foot	Microfibers were found in all organs, including organs which are not involved with ingestion. It indicates that the accumulation of microplastics is not only due to ingestion process, but also adherence to the surface of mussel tissues. The highest concentration of microfibers was found in gut, both after exposure (171 particles/g) and elimination (91 particles/g).	Kholandhasamy et al. (2018)
Mytilus galloprovincialis	PE and PS powders (virgin and contaminated with pyrene)	<100 µm	*Water*: 1.5 g L^{-1}; 7 days	Haemolymph, digestive glands, and gills	Localization of microplastics in haemolymph, gills and digestive tissues (and pyrene accumulation in the latter); alterations of immunological responses; lysosomal compartment; peroxisomal proliferation; antioxidant system, neurotoxic effects; onset of genotoxicity; changes in gene expression profile.	Avio et al. (2015)
Mytilus spp.	PS microbeads	2 and 6 µm	*Water*: 32 µg L^{-1} (alone) and 30 µg L^{-1} (combination withfluoranthene); 7 days exposure and 7 days depuration	Haemolymph, digestive glands, and gills	A higher fluoranthene concentration was detected in mussels exposed to fluoranthene in combination with micro-PS (after depuration); increased haemocyte mortality; triggered substantial modulation of cellular oxidative balance (increased reactive oxygen species production in haemocytes, and increased anti-oxidant and glutathione-related enzymes in mussel tissues).	Paul-Pont et al. (2016)
Scrobicularia plana (Clam)	PS spheres	20 µm	*Water*: 1 mg L^{-1}; 14 days followed by 7 days depuration	Haemolymph, gills and digestive glands	PS were not eliminated after 7 days depuration; influenced antioxidant capacity; DNA damage; induced neurotoxicity and oxidative damage	Kesy et al. (2016)

(*Continued*)

Table 5. (Continued).

	Species	Polymer type & shape	Size	Exposure route: concentration; duration	Endpoint(s)	Observed effect(s)	Reference
	Crassostrea gigas (Pacific oyster)	PS spheres	2 and 6 µm	*Water*: 0.023 mg L^{-1}; 2 months	Haemolymph, digestive glands, and gonads	Increased food consumption suggesting increased stress and energy demand to maintain homeostasis; reproductive disruption (significant decreases in oocyte number, diameter and sperm velocity); reduction of offspring performance during larval stages	Sussarellu et al. (2016)
	Carcinus maenas	PP fibres	<5 mm	*Dietary*: 0, 0.3, 0.6, 1%; 4 weeks	Faeces	Reduced food consumption and significant reduction of available energy for growth over 4 weeks in crabs fed with 1% plastic.	Watts et al. (2015)
	Uca rapax	PS fragments	180–250 µm	*Sediment*: 108 and 1000 mg kg^{-1} dry sediment; 2 months	gills, stomach,	hepatopancreas	Ingestion; PS detected in gills, stomach and hepatopancreas; PS in micro size can be translocated from stomach to hepatopancreas.
	Brennecke et al. (2015)		*Nephrops*	*norvegicus*	PE and PS microspheres (MS) loaded with PCBs	6 µm, 500–600 µm	*Dietary*: 155 mg microparticles (clean or loaded with PCBs) added to about 9.64 g of gelatin feed cubes. The concentration of PCB in each loaded microplastic was 1.35 µg. Exposure: 3 weeks
Tail tissues	Limited accumulation of PCBs residues in *Nephrops novergicus* tail tissues, when fed PCB-spiked PE MS of 500–600 µm. However, no accumulation was observed in case of exposure to PCB-spiked PS MS.	Devriese et al. (2017)					
	Atactodea striata	Microgranules PS	63–250 µm	*Water*: 0, 10, 1000 items/L; 10 days	Faeces and pseudofeces	Reduced clearance rate at high concentration of PS microgranules (more than two fold). As a consequence, it reduced the energy intake. The respiration rate and absorption efficiency remained stable. At high microplastic concentration, the number microplastics in faeces and pseudofaeces increased.	Xu et al. (2017)

(*Continued*)

Table 5. (Continued).

Species	Polymer type & shape	Size	Exposure route: concentration; duration	Endpoint(s)	Observed effect(s)	Reference
Daphia magna	PS-NH_2 Amino modified particles	52, 53, 57, 58, 120, 180 and 330 nm	*Water*: 0.025–0.150 g L^{-1}; 24 hours	The number of dead *D. magna* was counted every hour for 24 hours	Only the 52 nm sized particles showed concentration dependent toxicity. The animals were still alive after 24 hours exposed to 0.025 g L^{-1} polystyrene particles in water/algae medium. Above 0.075 g L^{-1} all animals were dead after 13 hours of exposure.	Mattsson et al. (2017)
Artemia franciscana	PS beads	0.1 µm	*Water*: 0.001, 0.01, 0.1, 1, 10 mg L^{-1}; 24 hours	Larvae	PS beads were accumulated in the gut of crustaceans after 24 and 48 hours- exposure. Some of the PS beads were excreted. No significant effect in mortality of larvae was observed. The exposure of PS beads in high concentration (>1 mg L^{-1}) for 48 hours changed swimming activity of crustaceans and indicated the induced of neurotoxic and oxidative stress.	Gambardella et al. (2017)
Palaemonetes pugio	PS, PE (spheres); PP (fragment and fibre)	Spheres: 165 µm; fragments and fibres (34 and 93 µm)	*Water*: 50,000 particles/L; 3 hours	Whole body, gut and respiratory chamber	Ingestion and ventilation are the main uptake pathways and the number of ingested particles is influenced by the shape (i.e. fragments > spheres > fibres). The residence time of microplastics in digestive tracts and gills ranged from 27 –75 hours and from 27–45 hours respectively. The uptake of microplastics resulted in acute toxicity for crustaceans, indicated by a mortality range from 0% – 55%. Particles in the form of 93 µm fibres showed the highest mortality rate.	Gray and Weinstein (2017)
Thylonella gemmata; *Holothuria floridana*; *Holothuria grisea*; *Cucumaria frondosa*	PVC fragment Nylon fragment PVC pellets	0.25–15 mm 0.25–1.5mm 4.0 mm	*Sediment*: 10 g PVC fragment/600 ml silica sand, 65 g PVC pellet/600 ml silica sand, 2 g nylon fragment/600 ml silica sand; 4 hours feeding. After the exposure, samples were moved to different tank for 20–25 hours.	faecal pellets, sand grains defecated	All specimen ingested plastic particles (PVC both fragments and pellets, as well as nylon fragments). The sea cucumbers ingested 1.4–20.5 times more PVC fragments than expected (the number of plastics pieces expected to be ingested for a given volume of sand), and ingested 2–138 fold more for nylon line. Two of 4 species can ingest PVC pellets	Graham and Thompson (2009)
Amphibalanus amphitrite	PS beads	0.1 µm	*Water*: 0.001, 0.01, 0.1, 1, 10 mg L^{-1}; 24–48 hours	Larvae	PS beads were accumulated in the gut of barnacles at very high concentration (≥ 1 mg L^{-1}) after 24 and 48 hours-exposure, and they did not excrete the particles. No significant effect in mortality of larvae was observed. The exposure of PS beads for 48 hours has induced of neurotoxic and oxidative stress.	Gambardella et al. (2017)

PE: polyethylene, HDPE: high density polyethylene, PP: polypropylene, PS: polystyrene, PVC: polyvinyl chloride

Standardization

Quality assurance protocols improve reproducibility and representativeness, but do not guarantee comparability of results between studies. The number of variables influencing microplastic measurements is endless. Examples include the mesh size of filters, sampling depth, tissues analysed, destruction techniques, analytical identification techniques and detection limits. The results in Tables 1–4 illustrate this enormous variability, with lower size limits varying from 4.3–1,400 µm (sediment) and 30–500 µm (water column), sampling depth of the water column varying from < 400 µm (surface microlayer) to 4.5 m, identification techniques covering visual inspection, FTIR and Raman spectroscopy, and tissues analysed covering the gastro-intestinal tract/guts/viscera, stomach/gut, gills, muscle/flesh and liver. The need for standardization is widely recognized, e.g. by international organisations such as GESAMP (2016). Standardization should not reduce scientific freedom by restricting the analysis to a limited set of parameters and techniques, but increase comparability between studies by the inclusion of a set of standardized parameters and techniques.

The need for standardization is most urgent for the units in which the amount of microplastics is being reported. Monitoring studies often report the number of particles in a medium without providing details about the mass or volume of these particles (Tables 1–4). Toxicity studies typically express exposure levels based on the mass of the particles (Table 5). Both units can be converted, provided particle characteristics (i.e., size, shape and material) are specified. However, determination of the size, shape and material of each individual particle is a time consuming and expensive effort which can be unfeasible in practice. As a bare minimum, monitoring studies should report the total weight and volume of the particles detected, next to the number of particles. This would allow concentrations of microplastics to be expressed in mg of plastics per unit of medium, enhancing the compatibility of results between monitoring and toxicity studies.

Microplastics in sediment, water, and seafood samples

Our results show that microplastics are widely distributed in the waters, sediments and seafood of coastal areas around the world (Tables 1–4). A formal statistical analysis of the spatial patterns in the data and the associations with pollution sources was not performed because of the low number and limited comparability of the data. Nonetheless, high levels of microplastics tend to occur in areas with high population densities, intensive recreation, intensive fisheries, and relatively poor waste management facilities, such as in China (Qiu et al. 2015), Canada (Desforges et al. 2014; Mathalon and Hill 2014), Mexico (Retama et al. 2016), and South Korea (Chae et al. 2015; Song et al. 2015). More data and analyses are needed to confirm these trends and to further explore potential associations with pollution sources and environmental factors such as currents and winds. This endeavour would greatly benefit from the creation of a global database on microplastics detected in water, sediments and organisms, including seafood. Such a global database would also be very useful for the identification of hotspots, and to monitor the impact of mitigation measures. Several non-profit organizations have compiled global data on microplastics (Stöhr 2016; Christiansen 2018), but these initiatives currently lack the scientific details required to explore the relationship between microplastics characteristics (e.g., size, material type, form) and particle behaviour, e.g. dispersion in water, uptake in organisms, trophic transfer and potential toxicity.

The present study reviewed microplastic contamination in a wide range of coastal seafood species (Tables 3 and 4). The number of microplastics ingested by seafood can be influenced by many factors, such as habitat and feeding behaviour. Our results show that pelagic fish ingest significantly more microplastics than fish from other habitats, which is in line with the results of Rummel et al. (2016) and Guven et al. (2017). Furthermore, our results indicate that fish from sites with a higher abundance of microplastics in water and sediment ingest a higher number of microplastic particles than fish from less polluted sites, which was also found by Li et al. (2016) and Guven et al. (2017). These results indicate that habitat type and the contamination level of the habitat are important determinants in microplastics exposure and accumulation. Feeding behaviour may also affect microplastics ingestion. Non-selective feeders (e.g. mackerels) and deposit

feeders (e.g. sea cucumbers) tend to ingest more microplastics than other seafood, which was also reported in previous studies (Graham and Thompson 2009; Rummel et al. 2016; Taylor et al. 2016).

Toxic effects in seafood species

Effects of microplastics in coastal fish and shellfish species have been demonstrated in a substantial number of laboratory experiments (Table 5). The effects are observed at all levels of biological organisation, i.e. molecular (e.g., gene expression, oxidative stress, DNA damage), cellular (histopathological alterations, immunological responses, haemocyte mortality, modulation of the oxidative balance), tissue (morphological changes, inflammation, neurotoxicity), functional (reproductive disruption, energy balance) and individual (mortality, behavioural changes). However, the available data do not allow the identification of clear cause and effect pathways. For example, increased gene expression may be triggered by microplastics directly, but it can also be an indirect response. It is even not clear whether the effects observed are the result of the additives and contaminants that leach from the particles, or of the physical impact of the particles themselves. Effects such as inflammation, immunological responses and oxidative stress are often observed after exposure to small particles and attributed to their physical impact (Wright and Kelly 2017), but these effects can also be triggered by chemical stressors (Jeng et al. 2011). Some studies show a clear dose- and time-dependency of the effects observed (Peda et al. 2016; Mattsson et al. 2017), but the establishment of relationships across multiple studies is hampered by the use of different particles (size, shape and material) and exposure regimes. More information on the relationship between microplastics characteristics and their behaviour in the environment is needed to determine which are the most relevant drivers of microplastic toxicity (Horton et al. 2017; Lambert et al. 2017).

Linking monitoring studies to toxicity experiments

An important risk management question is whether the toxic effects detected in laboratory studies also occur in the field. Since reliable data showing toxicity of microplastics under field conditions are lacking, this question can only be answered tentatively by linking the results of monitoring studies to those of toxicity experiments under laboratory conditions. As outlined above, this linkage is hampered by the use of different units. However, we tentatively converted the exposure levels reported in the toxicity studies (Table 5) into particle numbers per unit of medium, based on reported particle size, a specific mass of 1 g/ml for microplastics, and assuming particles are either spherical, cylindrical or block-shaped. The results are presented in Table 7.

For exposure through the water phase, the levels applied in the toxicity studies vary between 10^4–10^{17} particles/m^3. The water concentrations detected in the field are generally much lower, varying between 10^{-3}–10^5 particles/m^3 (Table 2), with the highest concentrations measured in subsurface microlayers. The lowest particle concentration at which adverse effects were detected was 7.46×10^6 particles/m^3 (von Moos et al. 2012) using grain-sized particles of 0–80 µm. This still is a factor of 50 higher than the highest concentration measured in the surface microlayer and a factor of 800 higher than the highest concentration measured in (sub)surface seawater.

In dietary studies, exposure levels range from 10^3–10^7 particles/g food (Table 5), with adverse effects starting at 2.44×10^6 particles/g food (Watts et al. 2015). The maximum concentrations found in human seafood (Table 6) are almost a factor of 100,000 lower than the minimum reported effect concentration. For sediment studies, the range in exposure levels used in toxicity experiments is small, i.e. $1.7–5.4 \times 10^5$ particles/kg sediment. Adverse effects were not reported at these levels (Graham and Thompson 2009; Brennecke et al. 2015). Detected environmental concentrations are at least a factor of 20 lower, varying between 0–10^4 particles/kg sediment (Table 1).

It can be concluded that the levels of microplastics detected in the environment tend to be much lower than the exposure levels applied in the laboratory and at which adverse effects in seafood species are being detected. Especially in dietary studies, exposure levels are much higher than the levels detected in the field. Indeed, many of the toxicity studies apply exposure levels a factor of 10^3–10^9 higher, triggering questions about the environmental relevance of these studies. Nevertheless, the application of very high exposure levels is a normal practice in

toxicity tests in order to establish the value of no observed adverse effect level (NOAEL) and to derive a margin of safety. In line with Lenz et al. (2016b), we recommend toxicity studies using exposure concentrations as close as possible to field levels, i.e. 10^{0}-10^{5} particles/m^3 for water, 10^{0}-10^{4} particles/kg for sediment and 10^{0}-10^{2} particles/g food for dietary studies (Tables 1,2 & 6). These numbers apply to particles in the size range of 20–5,000 µm.

Although the levels detected in the environment tend to be much lower than the effect levels from toxicity experiments, this does not automatically imply current environmental levels are safe for seafood species. One important source of uncertainty is the lower size limit at which particles are being detected in monitoring studies, which varies between 4.3 and 1,400 µm (Tables 1–2). Table 5 shows that particles much smaller than this detection limit, i.e. smaller than 1–10 µm, can already trigger adverse effects. Indeed, many studies tend to use very small size of plastic particles, even in nano sizes. The smallest size is 30 nm, and only one study used larger size microplastics (0.05–5 mm; Kholandhasamy et al. 2018). Although there are indications that the weathering of macroplastics can release very small particles (Lambert and Wagner 2016), their detection in environmental matrices remains a challenge and their fate is uncertain due to processes such as homo- and heteroaggregation (Olubukola et al. 2018). New methods and techniques are required to extract plastic particles smaller than 10 µm from environmental matrices.

Another factor limiting the field relevance of toxicity studies, is the limited representativeness of the particles used. Current experiments are often performed with particles of a single material, the same shape and a limited size range, whereas microplastics detected in environmental samples cover a variety of materials, shapes and sizes. Spheres, beads and pellets are most commonly used in toxicity tests (Table 5), whereas field particles are irregularly shaped as the majority results from the degradation of macroplastics. PP, PE, PS, PET, PA, and PL were the most frequently reported microplastic types in both environmental and seafood samples (Tables 1–4). This finding is in line with the fact that PP, PE (including HDPE and LDPE), PET and PS are among the six most produced plastics globally (PlasticsEurope 2017). However, toxicity experiments with particles of divergent composition are lacking. One option to increase the field relevance of laboratory toxicity studies is to perform the experiments with microplastic particles extracted from field samples, although it will be a challenge to extract these particles in sufficient quantities and in a representative manner.

Human health risks

The widespread presence of microplastics in coastal seafood species entails a potential human health risk, although adverse effects at estimated exposure levels (Table 6) seem highly unlikely. Several attempts to assess the risks associated with the presence of microplastics in seafood have been initiated (Brennecke et al. 2015), but most of these are related to the transfer of toxic organic pollutants. A human health risk assessment exclusively based on the effects of microplastics is still lacking.

Quantitative risk assessment is hampered by limited data on the fate and effects of microplastics in humans and related species. Small size microplastics and nano-sized plastics (<150 µm) have been shown to translocate across the gut epithelium in humans and rodents, resulting in systemic exposure (Browne et al. 2008). Translocation of even larger plastic particles (up to 438 µm) has been demonstrated in fish (FAO 2017a). However, quantification of the amount of particles being translocated remains a challenge due to lacking data, particularly with regard to degradation and excretion. Based on data obtained from previous studies on the absorption of microparticles in mammals, EFSA (2016) predicts the intestinal absorption of microplastics in humans to be limited (≤ 0.3%). Particle size, composition, surface charge and hydrophilicity are probably the most important factors influencing intestinal absorption (Hussain et al. 2001; Della Torre et al. 2014; EFSA 2016).

Toxicity testing with human cell lines can support human health risk assessment. An example is the study of Schirinzi et al. (2017), who conducted an *in vitro* study exposing cerebral and epithelial human cells (T98G and HeLa, respectively) to PE and PS microspheres. Exposure did not lead to significant reduction of cell viability (i.e., no cytolysis occurred), but the generation of reactive oxygen species (ROS) was induced at a concentration of 10 µg mL^{-1};

Table 6. Estimated intake of microplastics through seafood consumption for different regions in the world.

Seafood	Region	Seafood consumption (kg/capita/year)[a]	Concentration of microplastics (# particles/g)[b]	Estimation of microplastic intake (particle/capita/year)	
				Minimum	Maximum
Marine fish	America	0.53	0.02–25.9	11	13,727
	Oceania	2.53	NA*	NA*	NA*
	Asia	1.62	0.57–1.85	923	2,997
	Europe	0.53	0.06–2.00	32	1,060
	Africa	0.87	NA*	NA*	NA*
	Global	*1.25*	*0.02–25.9*[c]	*25*	*32,375*
Crustacean	America	2.54	NA*	NA*	NA*
	Oceania	3.77	NA*	NA*	NA*
	Asia	1.99	NA*	NA*	NA*
	Europe	1.76	0.18–10.9	317	19,184
	Africa	0.15	NA*	NA*	NA*
	Global	*1.79*	*0.18–10.9* [c]	*322*	*19,511*
Molluscs	America	1.64	NA*	NA*	NA*
	Oceania	2.65	NA*	NA*	NA*
	Asia	3.37	0.9–10.5	3,033	**35,385**
	Europe	1.95	0.2–13.1	390	25,545
	Africa	0.03	NA*	NA*	NA*
	Global	*2.5*	*0.2–13.1* [c]	*500*	*32,750*

[a] Based on seafood consumption data in 2013 from FAO (FAO 2017a)
[b] Taken from Tables 3 and 4, the minimum and maximum concentration found in each group from each region
[c] Taken from Tables 3 and 4, the minimum and maximum concentration found in each group from all regions
* NA (not available) if the microplastic concentration data were not reported in respective regions

indicative of oxidative stress. More studies using human cells exposed to different types and concentrations of microplastics are required for further investigation, especially to determine the type of microplastics that induce toxicity and the concentration at which adverse effects emerge.

Another challenge in conducting quantitative risk assessment of microplastics is the difficulty to carry out an accurate exposure assessment on fish consumption. As described in Table 3, microplastic particles are mostly found in the GI tract or in the stomach of fish. Fish innards are usually discarded during processing and may not directly pose a risk for human health, except for small fish (e.g. anchovies), shellfish such as bivalves and small shrimp that are usually eaten whole. Up till now, only two studies found microplastics in the edible part of fish (Akhbarizadeh et al. 2017a; Karami et al. 2017). However, it must be noted that the detection of microplastic, and in particular nanoplastics in seafood and other foodstuffs, is still in its infancy. Therefore, it can very well be that the currently predicted exposure levels are an underestimation.

Since viscera of fish and bones are used as ingredients for animal feeds (e.g. feed for poultry and fish), indirect human exposure to microplastics via the animal feed route is very likely (Bouwmeester et al. 2015; Rainieri and Barranco 2019) and requires further research.

Economic impacts

Microplastics may impact seafood stocks, as the particles can be ingested by small organisms at the bottom of the food chain, transferred to higher trophic levels, and can cause a decrease in fish harvest and quality (Van der Meulen et al. 2014; United Nations Environment Programme 2017). The observed toxic effects in fish and shellfish, such as reproductive disruption, reduction of energy for growth, and alteration in hunting and eating behaviour (Table 5), may ultimately result in reduced stocks of important commercial seafood species. The potential economic damage is substantial. A study by Van der Meulen et al. (2014) predicted that microplastics may lead to an annual income loss up to 0.7% every year for UK aquaculture. Assuming this figure applies to the global fishery and aquaculture industry, this would entail a loss of 1.18 million tonnes or 1.95 billon USD in 2015 (FAO 2017b).

Besides the direct effects of microplastics on seafood stocks, the indirect economic impact may even be larger. The lack of data on the presence and toxicity of microplastics creates uncertainty on whether microplastics in seafood can be considered safe or not. Although exposure levels for humans are low and adverse effects highly unlikely, laypeople are getting increasingly worried because of newspaper articles about the detection of microplastics in seafood.

Table 7. Estimated particle concentrations (final column) of the lowest exposure levels applied in the toxicity experiments as listed in Table 5.

Study	Size[a]	Shape	Lowest Exposure	Particle concentration
Exposure through the water phase				*#/m³ water*
Ferreira et al. (2016b)	1–5 µm	Spheres	0.184 mg L^{-1}	$1.30 \cdot 10^{10}$
Mattsson et al. (2017)	53 nm	Particles[1]	0.029 g L^{-1}	$3.72 \cdot 10^{17}$
Pitt et al. (2017)	51 nm	Particles[1]	0.1 mg L^{-1}	$1.44 \cdot 10^{15}$
Wegner et al. (2012)	30 nm	Spheres	0.1 mg mL^{-1}	$7.07 \cdot 10^{15}$
Van Cauwenberghe et al. (2015)	10, 30 & 90 µm	Spheres	110 #/mL	$1.10 \cdot 10^{8}$
Browne et al. (2008)	3 & 9.6 µm	Spheres	0.51 mg mL^{-1}	$1.10–36.1 \cdot 10^{9}$
von Moos et al. (2012)	0–80 µm	Grain[1]	2.5 mg mL^{-1}	7.46E+06
Kholandhasamy et al. (2018)	0.05–5 mm	Fibers[2]	2,000 #/L	$2.00 \cdot 10^{6}$
Avio et al. (2015)	<100 µm	Powder[1]	1.5 g L^{-1}	$2.29 \cdot 10^{10}$
Paul-Pont et al. (2016)	2 & 6 µm	Beads[1]	30 µg L^{-1}	$2.65–71.6 \cdot 10^{8}$
Kesy et al. (2016)	20 µm	Spheres	1 mg L^{-1}	$2.39 \cdot 10^{8}$
Sussarellu et al. (2016)	2 & 6 µm	Spheres	0.023 mg L^{-1}	$5.49–20.3 \cdot 10^{9}$
Xu et al. (2017)	63–250 nm	Granules[1]	10 #/L	$1.00 \cdot 10^{4}$
Mattsson et al. (2017)	52 nm	Particles[1]	0.025 g L^{-1}	$3.40 \cdot 10^{17}$
Mattsson et al. (2017)	53 nm	Particles[1]	0.025 g L^{-1}	$3.21 \cdot 10^{17}$
Mattsson et al. (2017)	57 nm	Particles[1]	0.025 g L^{-1}	$2.58 \cdot 10^{17}$
Mattsson et al. (2017)	58 nm	Particles[1]	0.025 g L^{-1}	$2.45 \cdot 10^{17}$
Mattsson et al. (2017)	120 nm	Particles[1]	0.025 g L^{-1}	$2.76 \cdot 10^{16}$
Mattsson et al. (2017)	180 nm	Particles[1]	0.025 g L^{-1}	$8.19 \cdot 10^{15}$
Mattsson et al. (2017)	330 nm	Particles[1]	0.025 g L^{-1}	$1.33 \cdot 10^{15}$
Gambardella et al. (2017)	0.1 µm	Beads[1]	0.001 mg L^{-1}	$1.91 \cdot 10^{12}$
Gray and Weinstein (2017)	165 µm	Spheres	50,000 #/L	$5.00 \cdot 10^{7}$
Gambardella et al. (2017)	0.1 µm	Beads[1]	0.001 mg L^{-1}	$1.91 \cdot 10^{12}$
Dietary exposure				*#/g food*
Mazurais et al. (2015)	10–45 µm	Beads[1]	0.12% (w/w)	$1.10 \cdot 10^{7}$
Peda et al. (2016)	<0.3 mm	Unknown[1]	0.1% (w/w)	$5.66 \cdot 10^{4}$
Rochman et al. (2014)	<0.5 mm	Pellets[3]	10% (w/w)	$1.22 \cdot 10^{6}$
Watts et al. (2015)	<5 mm	Fibres[2]	0.3%	$2.44 \cdot 10^{5}$
Devriese et al. (2017)	6 µm	Spheres	155 mg/9.64 g cube (= 15.9 mg g^{-1})	$1.42 \cdot 10^{8}$
Devriese et al. (2017)	500–600 µm	Spheres	155 mg/9.64 g cube (= 15.9 mg g^{-1})	$1.85 \cdot 10^{2}$
Exposure through sediment				*#/kg sediment*
Brennecke et al. (2015)	180–250 µm	Fragments[4]	108 mg kg^{-1}	$5.43 \cdot 10^{5}$
Graham and Thompson (2009)	0.25–1.5 mm	Fragments[4]	2 g/600 ml sand[5]	$1.66 \cdot 10^{5}$

[a] For spherical particles, size is assumed to represent the diameter; for all other shapes, size is assumed to represent length. If '<' was used, half the maximum value was used for length/diameter; [1] Particles, grains, powders, beads, granules and unknown shapes were assumed spherical; [2] Fibres were assumed to be cylindrical with a 1:100 diameter to length ratio; 3 Pellets were assumed to be cylindrical with a 1:5 diameter to length ratio; [4] Fragments were assumed to be block-shaped with 10:2:1 ratio for length: width: height; [5] For sand, a specific weight of 1.5g mL^{-1} was assumed.

As scientists, we can tell people not to worry, but this is a vulnerable strategy. Critics will argue microplastics do not belong in seafood and will point towards lacking exposure and toxicological data, implying we cannot guarantee the safety of our seafood. The raising awareness of microplastics contamination in seafood may thus lead to the less seafood consumption. Stakes are high since the global seafood market is a serious economic factor.

Management options

Considering the stakes involved, policy makers have to operate carefully and wisely when dealing with microplastics in seafood. Management options are manifold; each with their own pros and cons. Examples include a reduction in the use of plastics, improved waste management, emission reduction, setting food safety standards and dietary restrictions. Preventive measures tackle the problem at its source, but it takes a long time before such measures result in reduced pollution levels. Dietary restrictions have immediate effect, but may seriously harm the seafood sector. Since microplastics are commonly found in the digestive tracts of seafood, reducing the intake of seafood that is eaten whole (e.g. small fishes like anchovy, edible bivalves, small shrimp), foods prepared with fish viscera, or products from animals raised with fish viscera, will immediately reduce microplastics intake. However, considering the potentially enormous economic impacts, dietary restrictions seem premature as long as toxic effects at current exposure levels have not been demonstrated.

A more realistic option is to set a provisional action level based upon a predetermined percentile,

e.g. the 95th percentile, of current detected levels, especially for small fish and shellfish. Action levels are widely used by the United States Food & Drug Administration (USFDA 2000), e.g. for contaminants that do not belong in food, but cannot be avoided. These levels are not based on direct health effects but on aesthetic considerations, e.g. the presence of natural or unavoidable defects in food such as insect larvae and eggs (USFDA 2005). Setting an action level for microplastics in seafood would have some important benefits:

(1) it will oblige food safety managers to regularly check the presence of microplastics in seafood, resulting in a valuable source of information on current exposure levels;
(2) it gives a clear signal to the general public that food safety managers take safety seriously and that they are 'on top of it', i.e. by carefully monitoring the developments, allowing interventions if deemed necessary;
(3) it will give a strong signal to all stakeholders involved in plastic (waste) management, i.e. that plastic pollution in oceans should be prevented because it is potentially threatening a valuable resource, i.e., seafood.

The establishment of a provisional action level for seafood will also have less favourable consequences, particularly economically. It requires regular monitoring of microplastics in seafood with standardized measurement protocols and robust quality control procedures. And if an action level is being exceeded, action should be taken which may vary from long-term remediation options to removing the product from the market (USFDA 2000). The implementation of an action level and determining its numerical value therefore requires careful consideration and requires input from all stakeholders involved.

Conclusions

The presence of microplastics in coastal areas worldwide is significant and raises serious concerns due to their ability to transfer to coastal seafood. Commercial fish and shellfish in coastal areas have been shown to be contaminated by a wide array of microplastics in terms of types, shapes and sizes. The concentrations found in seafood tend to correlate with the pollution levels of the environment. It has, furthermore, been demonstrated that microplastics can cause different adverse effects in seafood species and mortality of fish larvae, although the exposure levels were generally orders of magnitude higher than those encountered in the water column and sediments of coastal areas. It is concluded that current data availability is insufficient to perform an adequate risk assessment for microplastics affecting seafood species and human health. Further research is needed that relates the presence of microplastics to potential toxic effects on seafood species and human health, e.g. by using a combination of laboratory experiments with seafood species and *in vitro* assays with human cell lines. The microplastics tested should be similar in shape and size as the ones found in field samples. For now, the presence of microplastics in coastal seafood can be considered a form of unintentional food contamination. Considering the importance of assuring seafood quality and safety, and in the absence of a robust risk-based approach, food safety managers may consider to set a provisional action level for microplastics in seafood. This will be a strong impetus for gathering new data and is a clear signal that microplastics do not belong in seafood.

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