

A Review of Microplastic Identification and Characterization Methods in Aquatic Environments

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Abstract

Plastic waste has become a major global issue, with over 390.7 million tons of plastic produced in 2021. Because of its durability, low recycling rates, poor waste management, and maritime use, a considerable portion of plastic waste ends up in aquatic environments. Photo-oxidation and other mechanisms degrade plastics into microplastics (MPs), which are particles smaller than 5 mm. MPs can spread through the aerial, terrestrial, and aquatic areas, and running waterways serve as conduits for MP transport across various ecosystems. MPs have been found at various levels of the food web, and animals can ingest, inhale, or absorb them through their skin. MPs pose a significant health risk to flora and fauna, including marine creatures and humans, due to their small size, diverse colors, high abundance, and ability to adsorb antibiotic-resistant pathogens, causing cytotoxicity, acute reactions, undesirable immunological responses, neurotoxicity, and DNA alteration. MPs have a negative economic impact on industries such as agriculture, fishing, tourism, etc. Detecting and quantifying the presence of MPs is therefore critical. The purpose of this paper is to provide an overview of the various techniques and equipment used to detect and characterize MPs in aqueous environments. Identifying and educating the public about the primary sources of plastic pollution can help reduce the number of MPs in the environment.

Keywords

microplastics, microplastic identification, microplastic characterization, aquatic environment, plastic pollution

1 Introduction

Plastics encompass a wide range of synthetic or semi-synthetic materials derived from oil, coal, and chemical compounds. The global production of plastic has been consistently increasing, surpassing 300 million tons annually since 2014 and reaching a peak of 390.7 million tons in 2021 [1–4]. These versatile materials find applications in various sectors worldwide, including packaging (146 million metric tonnes, 36%), construction industry (65 million metric tonnes, 16%), textile (47 million metric tonnes, 12%), consumer products (e.g., toys, toothbrushes: 42 million metric tonnes, 10%), transportation (e.g., headlights: 27 million metric tonnes, 7%), electronics (18 million metric tonnes, 4%), industrial machinery (3 million metric tonnes, 1%), and other sectors (59 million metric tonnes, 14%) [5]. A significant proportion of the plastics produced globally enter the environment and marine ecosystems through various pathways, leading to long-term persistence. Their non-biodegradable nature, prolonged durability, low recycling rates, and inadequate waste

management contribute to this issue [1, 6–13]. Plastics can degrade into smaller fragments through processes such as photooxidation (UV radiation), physical stress, temperature fluctuations, wave action, salinity variations in marine environments, and microbial degradation, among others [1, 2, 6–11]. Based on their size, plastic litter can be categorized as follows: Megaplastics (>1 m), Macroplastics (1 m–2.5 cm), Mesoplastics (2.5 mm–5 mm), Microplastics (<5 mm–1 μ m), Nanoplastics (<1 μ m), and Microbeads/Microexfoliates, which refer to small plastic granules used in personal care products like toothpaste and facial cleansers [1, 2, 6, 8, 9, 14].

In the realm of microplastic (MP) pollution, there exist two main categories: primary microplastics and secondary microplastics [9]. Primary microplastics are intentionally manufactured in sizes smaller than 5 mm for specific purposes, such as microbeads and plastic pellets utilized in the production of plastic goods. On the other hand, secondary microplastics originate from the

degradation and fragmentation of larger plastic materials present in the environment, resulting in the formation of synthetic fibers and plastic fragments [9, 15, 16].

By the Source-Pathway-Receptor model, microplastic (MP) particles originating from diverse sources permeate the aerial, aquatic systems encompassing marine, riverine, and lacustrine environments, as well as terrestrial ecosystems, even in polar ice through various pathways, including rivers, direct discharge into the ocean, atmospheric deposition, wind dispersion, wastewater and runoff from urban and industrial areas [17]. Subsequently, MP particles tend to accumulate in specific areas, such as oceanic regions, where they interact with receptors present in nature, including flora and fauna [17]. Receptors can be exposed to MP particles through ingestion, inhalation, and dermal absorption [17]. Moreover, MP particles have the potential to propagate through the food web, ultimately reaching humans. This transfer of MP particles across the food web can have implications for the health and well-being of various species [17].

Rivers play a pivotal role as the primary conduit for the transport of microplastics (MPs) from terrestrial environments to marine ecosystems, particularly in freshwater regions [17, 18]. The worldwide ocean is expected to contain 4.85 trillion MP particles, with up to 80 percent of them plausible to have been transmitted from river networks [19]; every year, rivers discharge between 1.15 and 2.41 million tons of plastic debris into the ocean, with approximately 74 percent of emissions happening between May and October [20]. The majority of global pollution can be attributed to the top 20 most polluted rivers, predominantly located in Asia, accounting for 67% of the total pollution [20]. The main sources of MP particle pollutants include the effluent from clothes washing processes (35%), tire abrasion (28%), construction industries (24%), road paint (7%), shipping activities and ship coatings (3.7%), cosmetics (2%), and plastic pellets used in plastic production industries (0.3%). Another significant source of MP fibers has emerged during the COVID-19 pandemic, resulting in the abandonment or improper disposal of approximately 89 million face masks in the natural environment [21]. In February 2021, a substantial quantity of these masks was discovered in the ocean near Hong Kong [21]. These masks are primarily composed of polymers such as polypropylene, polyurethane, polyacrylonitrile, polystyrene, polycarbonate, polyethylene, and polyester [21].

Microplastics have been identified in multiple environmental compartments, including the air, water, and various food sources consumed by individuals [22]. Notably,

seafood such as shellfish, mussels, and fish products, as well as commonly consumed items like bottled water, honey, sea salt, and contaminated food products, have been found to contain microplastic particles. Additionally, the presence of microplastics can be attributed to the leaching of monomers from food packaging materials and other related sources; however, the extent of absorption and retention is unknown [22].

MPs could account for up to 30% of the Great Pacific Garbage Patch, which is now polluting the world's oceans. Wetland habitats show a negative correlation between MPs concentration, vegetation cover, and stem density [17]. In an ice core from east Antarctica, discovered 96 microplastic particles consisting of 14 types of polymers. Indoor airborne microfiber concentrations were between 1.0 and 60.0 microfibers per cubic meter (33% of which were found to be microplastics) [17]. Detected 2,649 particles of airborne microplastic in 10 samples of street dust in Tehran street dust samples, with concentrations ranging from 83 particles to 605 particles or 10 particles per 30 g of street dust [23]. Various scientific investigations have revealed that an adult male engaged in light physical activity inhales an estimated daily intake of 272 microplastic particles. Microplastics in the air are produced by synthetic fibers, abrasion of materials (e.g., automotive tires, etc.), construction materials, household furnishings, landfills, automobile emissions, industrial products, and resuspension of microplastics in surfaces, waste incineration [24]. Microplastic particles were discovered in the placentas of unborn infants for the first time in December 2020 at Arizona State University [17].

Because of the MP's size, density, and color, they are mistaken for food by various aquatic species such as fish, zooplankton, etc. clogging their digestive processes and transmitting false feeding signals [9, 25]. Over 280 marine species, including key food chain participants, have been found with MPs in their stomachs and intestines (up to 10% of their body weight). Ingested MPs have been linked to brain damage, behavioral disorders, reduced feeding rates, less energy for growth, and lower reproduction in fish. MPs can act as an egg-laying surface for marine insects, potentially causing ecological harm and species overlap [9, 25]. The toxicity of MPs can arise from the plastic polymer itself, additives, or other substances that come into contact with them and this issue has various detrimental consequences, including immunotoxicity and neurotoxicity, as well as altering the DNA [5, 26, 27]. Physical problems, chemical effects, reduced health, ecosystem impacts, dissemination of pathogens, and transport of pollutants are

potential consequences. Microplastics can host microbial communities, transport synthetic organic compounds, and provide new colonization habitats, increasing species overlap [28–31]. Also, antibiotic-resistant pathogens attached to MPs can be rapidly transported, spreading diseases to new locations. Since small bits of plastic are more likely to be ingested by wildlife and have a higher surface area, they can transport chemicals to and from the marine environment, microplastic contamination may be more environmentally harmful than bigger pieces of plastic [22, 32, 33].

Microplastics are commonly utilized as carriers for medicinal substances, facilitating the targeted delivery of drugs to various tissues and layers within the human body [22]. The specific characteristics of microplastics, including their size, shape, composition, and hydrophobicity, play a critical role in determining their bio-distribution, transportation, and ultimate impact on the body [22]. It has been observed that microplastic particles smaller than 135 μm can easily traverse the human respiratory system, leading to prolonged irritation of the alveoli [22]. Additionally, particles smaller than 110 μm have the potential to enter the bloodstream, urine, and cerebrospinal fluid, and accumulate in various organs [22]. Moreover, microplastics smaller than 130 μm can access the lymphatic system, being transported or deposited in lymph nodes, while particles with sizes below 20 μm have demonstrated deposition in the liver, kidneys, and intestine [22]. MPs may bind to heavy metals or other chemical substances in the environment and act as a vector for delivering them into the body, and microplastics may operate as disease vectors [30, 34, 35]. MP exposures have some serious neurological and psychological effects on the human body such as endocrine disruption, brain disorders, sexual dysfunction, prostate cancer, sperm count decrease, in men; decreased maternal behavior, ovarian cancer, embryo cell liver damage, fetal risk of developing breast cancer, in female; neurotoxic response, sex-dependent brain restructuring, nasal cavity cancer, metabolic disease, estrogen agonist androgen antagonist, airway impaction, respiratory disease, lung cancer, cardiovascular disease, breast cancer, diabetes, long-bowel cancer, liver disease, bladder cancer, in both genders [22, 32].

Soil ecosystems are a significant sink of MP particles. Human activities, including agricultural practices, machinery degradation, littering, sewage sludge application, and plastic mulching, contribute to plastic pollution in terrestrial habitats [36, 37]. Soil erosion during heavy

rainfall can transport MPs into riverine systems, which can return to the land during floods [36, 37]. Exposing MPs causes biophysical soil response, reducing the seed's germination, decreases soil PH, alters the size distribution of water-stable soil aggregates, increases the soil-water evaporation, changes soil properties, decreases the soil bulk density, increases the soil porosity, changes the soil microbial composition, stunting the earthworms and terrestrial plants' growth [36, 37].

Plastics have a mixed impact on global warming and Plastics have been highlighted as a potential agent of global change [36, 38, 39]. Microplastics (MPs) can be transported globally by wind due to their small size and low density. Atmospheric aerosols, including MPs, influence Earth's climate by absorbing and dispersing radiation. Effective Radiative Forcing (ERF) is used to measure their climate effects, with most aerosols offsetting greenhouse gas warming except for soot (black carbon) which is low for MPs in comparison to the others [38]. The atmospheric chemistry and cloud reactions resulting from MPs are not well understood. Plastic production contributes significantly to greenhouse gas emissions, releasing 850 million tons of carbon dioxide (CO_2) into the atmosphere in 2019. However, plastics can also contribute to energy savings due to their lightweight and durable nature compared to glass or metal [36, 38, 39].

Given the negative impact of MPs on the environment, the comprehensive identification and characterization of microplastics (MPs) in the environment, particularly in aquatic ecosystems, are of paramount importance. These particles have substantial implications for the overall health and functioning of Earth's ecosystems. In the subsequent sections, we will delve into the various methods employed for the identification and analysis of MPs.

2 Methodology and material

In this paper, a comprehensive literature review about sampling and identification of microplastics in the aquatic environment and their ecological impacts was conducted by consulting relevant resources available in reputable databases such as Google Scholar, Elsevier, and Web of Science by using some keywords such as microplastics (10263 numbers of papers), microplastic identification, microplastic characterization, microplastic pollution.

The current methodologies employed for the collection, extraction, and quantification of microplastics (MPs) from aquatic environments are characterized by time-consuming and labor-intensive procedures. Moreover,

these methodologies necessitate the utilization of sophisticated techniques and costly analytical tools [40]. It is worth noting that the sampling process itself significantly contributes to the overall uncertainty associated with the analysis outcomes.

The hydrological characteristics of a water body, including factors such as water density, wind patterns, currents, waves, and tides, along with seasonal runoff, regional geography, river morphology, and riverine environmental conditions (such as river length, catchment size, and population density), as well as the prevailing meteorological conditions, and physicochemical properties of identified particles (such as shape, size, and density), greatly influence the fate and transport of microplastics (MPs) [19, 40–51]. Additionally, the erosion and deposition dynamics of the sediment bed surface play a significant role in determining the burial, settling, or erosion of MPs from the riverbed [19, 40–51]. Other factors to be considered include the number and locations of sampling sites, sampling frequency, precision and accuracy of measurements, available equipment, and economic considerations [19, 40, 41, 43–51]. These various elements, individually or in combination, influence the trajectory of microplastic litter within the catchment region [19, 40, 41, 43–51]. Therefore, before establishing routine monitoring of MPs, it is essential to thoroughly evaluate the spatio-temporal distribution and behavior of MPs in the water compartment, taking into account all of these factors and aspects [19, 40, 41, 43–51].

To identify and characterize microplastic (MP) particles in aquatic environments, a comprehensive field study should be implemented [19, 40, 52]. The first step involves defining the objective of the study and selecting the appropriate study area and sampling strategies. Specialized instruments will be utilized to collect samples from the designated regions and aquatic phases (water surface, water column, sediments, and the ingested part by biota (Fig. 1 - III) [19, 40, 52], ensuring proper handling and transportation to the laboratory. In the laboratory, the samples will undergo extraction and characterization processes using advanced techniques such as Fourier-transform infrared (FTIR) spectroscopy or Raman spectroscopy [19, 40, 52]. Before analysis, the samples will be prepared by sieving and subjected to digestion methods to remove any potential adsorbed components [19, 40, 52]. Moreover, density separation techniques will be employed to differentiate the MP particles from other particles

present in the samples. In this context, various sampling and analysis methods for studying these particles will be discussed in detail [19, 40, 52].

Due to their small size, low weight, and density (ranging from 0.9 to 1.5 g/cm³), microplastic (MP) particles tend to float on the surface of water or reside in the lower region of the thermocline in marine waters. To ensure effective sampling, two methods are recommended: stationary sampling and dynamic sampling, depending on the specific environmental compartment being targeted (Fig. 1 - IV) [19, 40, 52]. In **dynamic sampling**, some trawls with a mesh size of less than 20 µm are towed to the boat by a 50–70 m long rope with two flowmeters (one inside and one outside) for filtration efficiency monitoring. It is recommended that the speed of the boat should be 0.5 knots otherwise turbulence at the mouth part makes the trawl will be damaged also, and the vertical distribution of particles will be changed. For having spatial distribution it is also recommended to take at least three samples for each length and from different points to cover the cross-section of the river. The sampling should be repeated at various times for temporal changes (e.g., daily, seasonal) [18, 19, 40, 53–56]. **Stationary sampling** is appropriate for non-navigable smaller rivers, streams, and creeks, with variable water regimes. The nets must be positioned in the opposite direction of the river's flow of water, and throughout the sampling period, a weight is employed to ensure a constant and consistent submersion depth of the nets. The duration of the sampling depends on the river's flow and the rate at which the nets clog. To reduce temporal and spatial variability, sample replication is frequently recommended [18, 19, 40, 53–56].

2.1 Sampling methodology

There are three general sampling methods introduced as follows [57] (Fig. 1 - II).

2.1.1 Selective sampling

In situations where discernible items such as large microplastics (ranging from 1 to 5 mm) and plastic pellets are visibly present on the surface of sediments and can be easily differentiated from the surrounding matrix, researchers commonly employ tweezers to directly retrieve these microplastics. However, caution must be exercised when encountering debris that lacks distinctive shapes, such as irregular, rough, or angular features, as there is a considerable risk of inadvertently overlooking such particles [41, 58, 59].

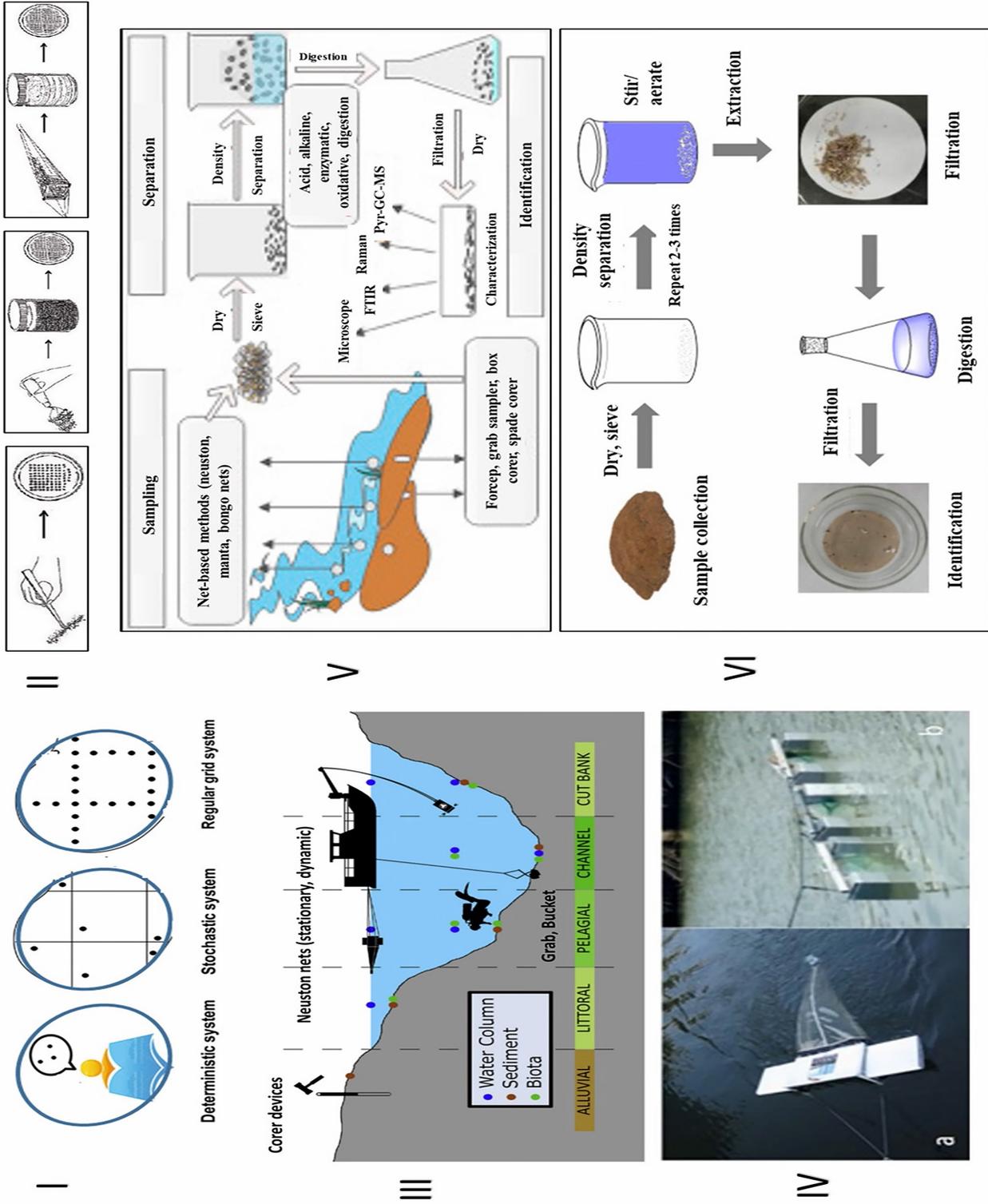


Fig. 1 I - Sampling strategies [19]; II - General sampling methods from left to right: Selective sampling, Bulk samples, Volume-reduced samples [57]; III - Sampling in the aqueous phase [55]; IV - a - Dynamic sampling, b - Stationary sampling [18]; V - Sampling process and preparation in the aquatic environment; VI - Sampling process and preparation in the soil environment [52]

2.1.2 Bulk samples

During the sampling process, a certain weight or volume of sediment is collected, not just microplastics. In the situation that MPs are too small, and they are not distinguishable by the naked eye, the abundance of particles is small, and filtration or sorting is needed after sampling, or when the particles are covered by sediments this method will be useful. Also, this method is less time-consuming, no special person will be required, and there are some standards for sampling. Further laboratory process is required after sampling [41, 58, 59].

2.1.3 Volume-reduced samples

Refers to samples where the volume of the bulk sample is reduced during sampling, preserving only the section of the sample that is of interest for future processing in both sediment and seawater samples. Samples of sedimentary environments can be sieved directly on the beach or board the boat, whereas volume-reduced samples of seawater are often produced by filtering huge amounts of water using nets. As the same as the bulk sampling further laboratory process is required after sampling [41, 58, 59].

2.2 Sampling process

2.2.1 Sampling strategies

Sampling will be performed in three types of strategies introduced as follows (Fig. 1 - I) [19, 60, 61].

Deterministic system: Refers to a sampling strategy based on existing knowledge and specific places [19, 60, 61].

Stochastic system: Produces a random pattern of equal subareas across the sampling region and statistically selects which ones will be sampled. Because in the case of MP, the transport processes in rivers are unknown this way may not be practical just yet, but it is strongly suggested when the statistical data on MP results or the cost/benefit ratio of a campaign needs to be assessed for future investigations [19, 60–62].

Regular system: By selecting sampling locations randomly or deterministically using a regular pattern, this technique decreases unknown sources of variance depending on limited time, financial, and expert staff capacity for MP sample processing, separation, analysis, and data interpretation, the number of samples and replicate, depth of sampling, the distance between sample locations might be changed (Single, duplicates, triplicates, quadruplicates, quintuplicates). Based on MSDF suggestion sampling should be performed in the top 5 cm, with a minimum of 5 replicates, at least 5 m apart [19, 60, 61].

2.2.2 Sampling instruments

The sampling methods and instruments will vary depending on the aqueous phase (surface water, water column, and sediment), the available equipment (Table 1), the aim of the study, and the matrix that must be collected; (Fig. 1 - V, VI) [18, 52].

Water surface sampling: Due to the low concentrations of MPs in the environment, sampling on the water surface usually necessitates water filtration or the collection of large sample volumes using volume-reduced or non-discrete sampling devices mostly Neuston, Plankton, and Manta nets of various mesh sizes (mesh size of 300–500 μm based on NOAA's size recommendations) [52]. Detection of samples up to 1 μm in size is possible. The considered devices for water surface sampling are listed in Table 1 [40, 61]. Additionally, bulk samples can be collected using discrete sampling devices such as Niskin bottles, Rosettes, Integrated Water Sampler (IWS), Buckets, Bottles, and Steel Samplers; KC Denmark A/S Research equipment, Silkeborg, Denmark, Auto-samplers [16, 18, 44, 45, 57, 63–70].

Water column sampling: Only a small fraction of MPs may concentrate in the water column due to their low density (most of them float on the water surface), and several factors can affect the MPs' vertical distribution which is measured with plankton nets or bongo nets [1, 18, 49, 52, 57, 71, 72]. The MPs' distribution throughout the water column depends on current conditions, flow turbulence, density and shape of particles, the density of the environment (freshwater density is lower than that of marine water), the adsorption of a biofilm, which raises the density of MPs and accelerates the sedimentation process [1, 49, 52, 57, 72]. For horizontal sampling after securing a weight (about 10–20 kg) to the net, it can be carefully lowered to the maximum depth without contacting the bed and then trawled obliquely at a rate of fewer than 2 knots to allow water to flow through the net in a steady stream. In vertical sampling, the net is raised to a certain depth and then lowered, sampling the whole water column [1, 18, 52, 57, 71]. Bongo nets have the benefit of sampling at various depths and locating MPs of various densities throughout the water column. After the sampling, the net should be washed to ensure that the whole particles accumulate in the sampling tube at the end of the net [1, 18, 52, 57, 71].

Furthermore, a recently developed custom-built apparatus has been devised and supplied for the quantification of microplastic (MP) particles distributed across different vertical profiles of the Austrian Danube River [70]. This

Table 1 Surface waters and sediment sampling devices [40, 61]

Sampling Device	Advantages	Disadvantages	Other information		
Surface water sampling devices	Manta net	A sampling of large volumes of water; The lateral wings allow the floating of the device and the sampling of the water surface.	Expensive equipment; Requires boat; The lower limit of detection is 333 µm; Clogging problems; Risk of sample contamination; Underestimation of the total buoyant microplastic amounts	15–240 min Non-discrete sampling devices	
	Neuston net	sampling large volumes of water; Widely used (useful for comparing positions). Plankton	Expensive equipment; Requires a boat; The lower limit of detection is 333 µm; Clogging problems; Risk of sample contamination; Underestimation of the total buoyant microplastic amounts. Expensive	30 min Non-discrete sampling devices	
	Plankton net	The lower limit of detection is 100 µm; Sampling of medium volumes of water; Possibility to sample the water column.	Expensive equipment; Requires a boat; Clogging problems; Sampling of lower volumes of water compared to Manta trawl; Risk of sample contamination; Underestimation of the total buoyant microplastic amounts.	30 min Non-discrete sampling devices	
	MP traps	Possibility to sample in several points of the water stream; Possibility to choose mesh dimensions from 100 µm to 333 µm.	Expensive equipment; May involve difficulty in anchoring to the riverbed; In the presence of a low flow rate, samples of the first 15 cm of water; Risk of contamination. Costly	30 min Non-discrete sampling devices	
	Autosampler	A well-known and precise volume of filtered water; Minimizes the risk of contamination; Allows a dimensional separation of the particles directly in the field	Costly equipment; Difficult and heavy to transport and deploy; Maybe very fragile; Requires electric energy; Requires a large amount of instrumentation	Non-discrete sampling devices	
	Pumping system	Allows the user to sample smaller MPs and fiber loss is limited; Well-known and precise volume of filtered water; Allows standardization of sampling. Relatively	Sampling a small volume of water; Requires energy to work and requires a boat; It can be challenging to transport and apply. Allows the sampling of a single point; Requires the transport of bulky samples to the lab; Sampling is less representative; Risk of sample contamination reactions integrated requires	15–180 min Non-discrete sampling devices Discrete	
	Niskin bottles/Jars/ Bottles/ Buckets/ Rosette/ Integrated water sampler (IWS)/ Ruttner bottles/ Friedinger bottles/ Bernatowicz bottles	Relatively quick and straightforward to use. Rosette provides multi-point measurements; Allows sampling at different depths; Allows the user to sample smaller MPs and fiber loss is limited; Well-known and precise volume of filtered water; Allows standardization of sampling.	Requires boat; Rosette can be challenging to transport; Sampling of a small volume of water; Maybe very fragile; Requires the transport of bulky samples to the lab; Sampling is less representative; Risk of sample contamination.	15–30 min Discrete sampling devices	
	Stainless-steel sieves/ Rotating Drum Sampler	Does not require specialized equipment; Quick and straightforward to use; Well-known and precise volume of filtered water; Allows choice of mesh size; Allows a dimensional separation of the particles directly in the field	A sampling of medium/low volumes of water; Requires the transport of a significant volume of water to the lab; Manual transfer of water with buckets; Potential contamination by the apparatus.	Timing depends on the mesh size Devices for surface microlayer	
	Sediment sampling devices	Dredges (Irish; Naturalist; Fast)	Large areas of the riverbed can be covered	Expensive, due to a boat being a hard requirement; No collection of a defined volume or depth of the sediment layer is possible; Sediment material is disturbed by overlaying water; Not feasible for onshore sampling	The device that is dragged across the riverbed and digs into the sediment from a boat
		Grab samplers (Ekman; Ekman-Birge; Forest Petersen; Franklin-Anderson; Lenz; Metabolizer; Ponar; Shipek; Dietz Lafond)	Large volumes can be collected; Simple handheld devices are available that can be used for MP onshore sampling	unpredictable penetration, which depends on the sediment composition; Perturbation of riverbed due to the impact and the subsequent opening and closing of jaws when the sample is recovered; Not applicable on hard surfaces or coarse sediments	The device contains two metallic jaws that close either automatically or by sending a mechanical, acoustical, or electric signal

Table 1 Surface waters and sediment sampling devices [40, 61] (continued)

Sampling Device	Advantages	Disadvantages	Other information
Sediment sampling devices Corers (Hand corers; Piston corers; Box corers; Gravity corers)	Collection of discrete samples at the defined point and depth; Various designs especially suited for working in different environments available; Useful when sampling on hard surfaces; Simple handheld devices available which can be used for MP onshore sampling	Disturbance and distortion of sediment if the tube is inserted into the sediment at a high velocity; Can be limited by freeze coring	The device contains two metallic jaws that close either automatically or by sending a mechanical, acoustical, or electric signal. The tube that is inserted into the sediment to collect a cylindrical sample

innovative system incorporates multiple nets of varying sizes, arranged in a stacked configuration one above the other, to effectively capture MPs in both medium and large streams of the river [70].

Sediment sampling: Silts, muds, and clays are found in low current velocity locations (e.g., dams and deltas), as well as in all types of lakes, in coarse fractions. In any case, sampling any form of lithology from shallow freshwater sediments should be the same [18, 52, 73–82]. The abundance of MPs on riverbanks depends on the factors relating to the hydrological conditions, and the land (deposition/erosion bank area, vegetation abundance/lack, finer/coarser sediments) [18, 52, 73–82]. Onshore sampling is best done in the swash zone with at least six samples per site, where MPs tend to accumulate under the wave's action or underwater at various depths [18, 52, 73–82]. For a sampling of sediment in freshwater areas three methods are being considered, collecting the particles manually from the depth of 5–32 cm for an area from 0.04–0.09 m², using corer devices with 50 mm specific diameter, using grabs (Ekman or Van Veen) with 225 cm² known area which it can readily be used to refer to a specific amount of material, and it much preferable than manual sampling [18, 52, 73–82].

2.2.3 Sample result units

A certain volume or weight of sediments is always sampled and based on the goal, and then the adopted methodologies in the case of water column or surface sampling, the MPs concentration can be expressed as, the number of particles per area (km², m²); the number of particles per volume (m³); the mass of MPs (g) per area (km², m²); the mass of MPs (g) per volume (m³, l) [18, 52, 83]. The filtered volume is normally calculated by measuring the precise volume of water going through the net with a mechanical or electronic flow meter connected at the net aperture, and it is equal to a multiple of the area of the mouth of the net, several revolutions of the impeller recorded by the flow meter,

hydraulic pitch [18, 52, 83]. In the case of dynamic sampling, the volume of filtered water is equal to the multiple areas of the mouth of the net, the distance covered during the tow [18, 52, 83]. In the case of stationary sampling, the flow meter is omitted, and by multiplying the surface of the submerged portion of the net, the sample duration, and the average river velocity (m/s), the total volume of water filtered through the net may be calculated [18, 52, 83]. Also, the MPs' transport rate for the cross-section of the river is equal to multiplying the MPs concentration (g/m³ or no. particles/m³), and the flow velocity of the river (m/s) [18, 52, 83]. The result of sediment sampling is expressed as the number of MPs per area (km², m²); the number of MPs per volume (m³); the number of MPs per mass (kg of dry sediment); the mass of MPs (g) per area (km², the m²); the mass of MPs (g) in volume (m³, l) [18, 52, 83].

2.2.4 Samples storing

After sampling sediment samples from water bodies and the bed or shoreline to transfer the samples to the laboratory for further analysis, samples are retained in special containers such as stainless steel containers, aluminum foil bags, glass bottles, and sealed bags [52, 58, 63, 76, 84–87]; The samples are then stored in labeled glass jars and kept at 4 °C, or frozen (below –20 °C) in darkness refrigerated until they are brought to analyze [52, 58, 63, 76, 84–87]. Moreover, for preventing biological reactions due to the biofilm adsorbed to MPs various fixing solutions (70–80% ethanol, 4–5% formalin) can be applied [52, 58, 63, 76, 84–87].

2.3 Microplastic sample preparation

2.3.1 Sieving

Sieving is used as an initial extraction step to reduce sample volume for MP separation procedures or to remove clay and silt-sized particles to minimize sediment grain aggregation. The mesh sizes (4 μm–5000 μm, a lower sieve mesh to distinguish between MP and NP) and the number

of sieves (one sieve or sieve cascades (series of sieves) of two and three sieves) depend on the desired specific particle size selection range and size classes. The materials that remain in the sieve are collected (and sorted), whereas the materials that pass through are usually discarded. Based on studies there are a variety of MP sieving methods for sediments, which are restricted by the fineness of the sediment material and the mesh sizes available [18, 19, 52, 53, 55, 57, 88–90].

Dry sieving: This method is efficient for fine sediment in the range of 40 μm and 125 μm which can cause particle aggregation or electrostatic charging [18, 19, 52, 57].

Wet sieving: The sieves must be rinsed with water from a spray nozzle situated above the sieve until the water leaving the sieve and no longer solid particles stack at the sieve outlet, after which the remaining water must be collected, filtered, and analyzed. Fibers and other low-density polymer particles are at risk of being discarded unintentionally [18, 19, 52].

Air-jet-sieving: Particle size to 20 μm can be separated by the rotating stream of filtered air on the sieve [18, 19, 52, 57].

2.3.2 Drying samples

Before analysis, the samples should be dried to a constant weight (dry weight (MP/g) set to compare the real content of microplastics in sediment without being impacted by sample humidity [18, 19, 52].

Oven drying: The quickest and most popular approach for removing residual water from sediment samples is to use a high-temperature oven (50 $^{\circ}\text{C}$ –100 $^{\circ}\text{C}$) which may lead to deformation, break, melting, and material degradation of some polymers (e.g., polyester-based fibers, polyvinyl chloride, PVC) [18, 19, 52]. Therefore, according to studies to prevent physical damage to polymer and save time, a temperature of less than 60 $^{\circ}\text{C}$ can be a good choice. The most appropriate option for larger, frequent monitoring programs with many samples is oven drying [18, 19, 52].

Air drying: Because of the laboratory environment and aerial conditions (humidity), drying samples at room temperature (25 $^{\circ}\text{C}$) is considered a long drying period and an insufficient method for drying wet samples. The drying process can be incomplete and contain variable final water content, and there is also the possibility of contamination via airborne MP particles [18, 19, 52, 57].

Freeze-drying (lyophilization): To eliminate residual water from MP river sediment samples, this procedure is often easier to process. Ice crystals form within the

sediment matrix during freezing, creating a porous structure following sublimation, preventing sediment grain aggregation. This approach has several significant drawbacks, including higher operating costs and smaller sample capacity than traditional ovens; also, there is no information about the effect of this procedure on the chemical and physical changes in MP [18, 19, 52, 57, 91–94].

2.3.3 Sample separation

MPs should be separated from the other particles due to the many interfering compounds commonly (Table 2) found in sediment samples, which might impact the quantification and identification of microplastics.

Density separation: This method is especially essential for sediments to distinguish the microplastics from the rest of the sample because of the specific density of plastic particles (0.01 to 2.3 g/cm^3); (Table 3), which varies based on the type of polymer and the production process [40]. The basis of this method, according to [52, 55, 95, 96], is mixing the sample with a high-density salt solution or saturated solution (e.g., saturated NaCl solution; Sodium metatungstate (SPT); Calcium chloride (CaCl_2); Sodium iodide (NaI); Zinc chloride (ZnCl_2) solution; Potassium formate solution; Canola oil and filtered water; Chloride solution and Munich Plastic Sediment Separator (MPSS)), after that, shake the mixture for 30 seconds to 2 hours, depending on the size of the sediment sample, and then let it rest to settle

Table 2 Chemical reagents used for density separation [40]

Reagent name	Chemical formula	Water solubility (g/L)	Density (g/cm^3)	Toxicity
Sodium chloride	NaCl	358 at 20 $^{\circ}\text{C}$	1.0–1.2	Low
sodium tungstate dehydrate	$\text{Na}_2\text{WO}_4 \cdot 2\text{H}_2\text{O}$	742 at 25 $^{\circ}\text{C}$	1.40	Low
Sodium bromide	NaBr	905 at 20 $^{\circ}\text{C}$ 3100	1.37–1.40	Low
Sodium polytungstate	$3\text{Na}_2\text{WO}_4 \cdot 9\text{WO}_3 \cdot \text{H}_2\text{O}$	3100 at 20 $^{\circ}\text{C}$	1.40	Low
Zinc chloride Zinc	ZnCl_2	4320 at 20 $^{\circ}\text{C}$	1.6–1.8	High
Zinc bromide	ZnBr_2	4470 at 20 $^{\circ}\text{C}$	1.71	High
Sodium iodide	NaI	1793 at 20 $^{\circ}\text{C}$	1.80	High

Table 3 The most common types of polymers and their density [40]

Material	Abbreviation	Density (g/cm ³)
Polystyrene	PS	0.01–1.06
Polypropylene	PP	0.85–0.92
Low-density polyethylene	LDPE	0.89–0.93
High-density polyethylene	HDPE	0.94–0.98
Freshwater	-	1.00
Seawater	-	1.025
Polyamide, Nylon 6,6	PA, PA 6,6	1.12–1.15
Polycarbonate	PC	1.20–1.22
Polyurethane	PU	1.20–1.26
Polyethylene terephthalate	PET	1.38–1.41
Polyvinyl chloride	PVC	1.38–1.41
Polytetrafluoroethylen	PTFE	2.10–2.30

down the suspended particles (from 2 min up to 6 h) [18, 19, 52, 55, 57, 71, 95, 96]. This process causes sediment to settle quickly to the bottom and low-density particles, such as microplastics, to suspend or float to the solution's upper layer and be separated. Although organic matter and phyllosilicate minerals (e.g., clay minerals and micas) are observed floating alongside MP particles [18, 19, 52, 55, 57, 71, 95, 96]. The differences between salts are attributable to their ability to separate and extract MPs based on their density, as well as their costs and toxicity, for instance, Sodium chloride (NaCl density: 1.2 g cm⁻³) is nontoxic, cost-effective salt; Sodium tungstate dehydrate (Na₂WO₄·2H₂O density: 1.4 g cm⁻³) is ineffective in density separation of heavy polymers such as polycarbonate (PC), polyurethane (PU), Polyethylene terephthalate (PET), polyvinyl chloride (PVC), or polytetrafluoroethylene (PTFE); Sodium iodide (NaI), (low concentrations) is a high-density and quite expensive salt which is useful for separation of most polymers [18, 19, 52, 55, 57, 71, 95, 96]; Zinc chloride (ZnCl₂) is the most effective and the least expensive method but it is Highly dangerous corrosive substance so, in should be careful in using, and disposal, and recycling of this reagent are required; salts like ZnCl₂, NaI, and ZnBr₂ are effective for high polymer density separation but they are highly soluble in water, therefore, larger quantities are required respect to NaCl, Na₂WO₄·2H₂O, or NaBr. Due to the high loads of complex biological matrices are exist in aquatic samples, a large volume of the oxidative or acid solution, and vast amounts of salts are required for digestion [18, 19, 52, 55, 57, 71, 95, 96].

Munich Plastic Sediment Separator (MPSS), established by Imhof et al. (2013) [78]: The device can separate large-volume samples and can analyze up to 6 L of

samples in one run. Also, the appropriate recovery rate of large microplastic (5–1 mm) and small microplastic (<1 mm) is observed [18, 19, 52, 57].

Korona-Walzen-Scheider (KWS), recommended by Felsing et al. (2018) [89]: This device can separate large-volume microplastic samples with a volume of 20 cm × 15 cm × 20 cm in one run with appropriate separation efficiency for microplastics of various sizes, densities, forms, and ages. Even for 63 μm particles, it demonstrates a substantial superiority in the separation of tiny microplastics. Removing the organic matter on the surface of the MPs is still needed [18, 19, 52, 57, 89].

Optimizing pressurized fluid extraction (PFE), by Fuller and Gautam (2016) [97]: A widely used technique for the extraction of organic pollutants from multiple environmental matrices and the performance, which is not affected by particle size, MPs would be either partially emulsified or solubilized under optimal PFE conditions then microplastics can be extracted from environmental samples. This method's limitation is the size distribution, and the morphology of particles would be obliterated [18, 19, 52, 57].

2.3.4 Filtration

The separated suspended particles from the density separation section should pass through a filtration paper with a pore size of 1 to 1.6 μm and 2 μm which is aided by vacuum [18, 19, 52, 57, 71, 95].

2.3.5 Sample purification (digestion)

Organic matter is in various forms in environmental samples. Purification techniques can be performed directly on environmental matrices to eliminate organic, inorganic, and detritus that can be confused for microplastics during measurement [18, 19, 52, 57]. When single microplastics are studied for their polymeric matrix composition (e.g., by FT-IR and Raman spectroscopy) or when optical identification methods for MP are utilized, purification procedures are also required. There are three common Purification strategies [18, 19, 52, 57].

Chemical degradation: Oxidizing, acidic, alkaline agents or Fenton's reagents are among the chemical methods of purification. MP samples are exposed to different chemical compounds (Table 4) [40] such as a mixture of Oxidizing agents or Peroxide mixed (10%–30% hydrogen peroxide (H₂O₂) solution) with sulfuric acid (H₂SO₄) which is used in some studies at special temperature range 20 to 80 °C and for special duration 1 hour

Table 4 Different reagents used for digestion of organic fraction necessary for MPs extraction from complex environmental matrices [40]

Reagents	Method	Hazard	Advantages	Disadvantages
HNO ₃	20 mL of HNO ₃ (22.5 M), 2 h heating (~100 °C), hot filtration (~80 °C)	Oxidizer, corrosive Corrosive,	Efficient in organic digestion	Possible degradation of PS, PA, and PE, makes the plastic yellow
HCl	4 mL of HCl at 20%	Corrosive, acute toxicity	Efficient in organic digestion (82.6%) of complex matrices (clams)	Degradation of polymers
NaOH	20 mL of NaOH (10 M) at 60 °C for 24 h KOH	Corrosive	Digestion efficiency up to 90%, stimulated by the rise of molarity and temperature	Degradation of PET and PVC
KOH	20 mL of KOH (1 M) at 18–21 °C for two days	Corrosive, irritant	Good organic digestion efficiency Efficient method	Requires lots of time, degradation of some polymers such as cellulose acetate and some biodegradable plastics
H ₂ O ₂	20 mL H ₂ O ₂ at 30% plus 20 mL of FeSO ₄ · 7H ₂ O (0.05 M) at 70 °C in stirring	Corrosive, harmful	Efficient in organic digestion	High concentrations could degrade the polymers
Cellulase, lipase, chitinase, protease, proteinase-K	5 mL of Protease A-01 + 25 mL of Tris-HCl buffer, 1 mL of Lipase FE-01 + 25 mL of Tris-HCl buffer; 5 mL of Amylase TXL + 25 mL of NaOAc buffer, 1 mL of Cellulase TXL + 25 mL of NaOAc buffer; 1 mL of Chitinase + 25 mL of NaOAc buffer	No danger	Good inorganic and biological material digestion; does not affect the polymers	Expensive requires lots of time

to 7 days. Oxidizing compounds are known to remove organic matter [96, 98, 99]. From the findings observed this peroxide solution will affect the polymer (polyamide (PA), polycarbonate (PC), and polypropylene (PP) pellets) by size reduction, discoloration, or for some polymers exposure to H₂O₂ (30%, 60 °C, 70 °C), is seemed unaffected [95]. The degradation of nylon and color changing of PE treated by 35% H₂O₂ at 50 °C for 8 days was reported [100]. During peroxide oxidation, between the reaction time, and temperatures, a balance relation should exist. By increasing the temperature, the reaction time should be reduced, so in temperature, some polymers will be unstable [18, 19, 52, 55, 57, 95, 96, 100–106]. In the case of using acidic or alkaline agents, some components like potassium hydroxide (KOH), sodium hydroxide (NaOH), nitric acid (HNO₃), hydrochloric acid (HCl), sulfuric acid (H₂SO₄), or perchloric acid (HClO₄) are used. By use of a 10% potassium hydroxide (KOH) solution, for 24 hours at 60 °C, the removal efficiency range will be from 99.6% to 99.8%, and it does not affect all tested plastic particles except cellulose acetate (CA) [18, 19, 52, 55, 57, 95, 96, 100–106]. For optimizing the digestion, the use of combined diluted solutions of H₂SO₄ (sulfuric acid), HNO₃ (nitric acid), and HCl (hydrochloric acid) with 30% H₂O₂ treatment is recommended but might lead to degradation and melting [18, 19, 52, 55, 57, 95, 96, 100–106]. Solutions of H₂SO₄ (sulfuric acid) can destroy or damage

the morphology of microplastics; the hot form used HNO₃ (nitric acid) can increase the destruction of organic matter between 93% and 98%, and cause the degradation of some polymer types, especially polystyrene, polyamide, and polyethylene; HCl (hydrochloric acid) has low efficiency in removing organic matter at room temperature [18, 19, 52, 55, 57, 95, 96, 100–106]. In the case of Fenton's reagent (Fenton reaction) which is an efficient method for removing organic matters, an advanced oxidation process using H₂O₂ in the presence of a catalyst (Fe²⁺) that can degrade efficiently organic components that are usually difficult to degrade in H₂O₂ alone [101]. Studies' results indicated that Fe²⁺ initiates and catalysis the decomposition of H₂O₂, thus oxidation occurs more rapidly [101, 102]. Based on NOAA recommendations, a Mixture of 30% H₂O₂ and 0.05 M ferrous iron (Fe²⁺) was used for river sediment samples, but it should be considered that H₂O₂ leads to an increase in the polymer transparency and make it thinner. Also, at the temperature setting (70 °C) oxidative damage and plastic polymer structure (for PA-6,6 polymer) degradation were observed [18, 19, 52, 55, 57, 95, 96, 100–106].

Enzymatic degradation: Organic matter such as carbohydrates, proteins, and lipids can be eliminated using a variety of technical enzymes (Chitinase, Proteinase, Lipase, Amylase, Corollas, Trypsin, Papain, Collagenase, and Cellulose) [41, 107]. In comparison to chemical digestion methods, this method is known to be gentle, but it

takes longer and costs more, especially for samples that include a considerable quantity of organic matter to digest and so require large amounts of enzymes. Enzymatic-oxidative techniques (optimal universal enzymatic purification protocol (UEPP)) were developed, which use a multi-enzyme approach to break down plankton, biota, and a variety of chemical substances while also releasing H₂O₂ [105, 108]. Although the techniques were rather efficient for aquatic samples, they still needed several days to remove enough organic material. To act at full capacity, enzymes require specific in-vitro conditions (concentration, temperature, and pH), and polymers that are sensitive to certain PH settings may be affected [104, 105, 109].

Mechanical treatment: Removing excess organic matrix from MP particles by using Ultrasonic cleaning. The brittle and aged artificial secondary MP particles were destroyed during ultrasonic use, and it can affect the size distribution analysis of these particles. The investigations showed that digestion efficiency in MP sewage sludge samples increases with ultra-sonication. This method is no more effective than using alkaline treatment alone [9, 18, 19, 52, 57, 104, 110].

2.4 Analytical measurements

After density separation and purification, the microplastic particles must be identified and quantified by the naked eye or by combination techniques of chemical-based analysis, and optical and spectroscopic techniques; (Table 5) [40].

2.4.1 MPs particle identification

Visual inspection: After cleaning the MP surfaces from organic and inorganic biofilms that attach them, visual examination is conducted under microscopes, and directly on the filter paper based on the physical characteristics of particles such as shape and color [18, 19, 52, 57]. Different microscopes could be used such as optical microscope (binocular biological microscope, dissecting microscope, and fluorescent microscope); and electron microscope (scanning electron microscope) [18, 19, 52, 57]. This method is open to bias and misjudgment due to MPs being hardly distinguishable from organic matter like quartz particles, animal parts, or small plant pieces. MPs should show a uniform homogeneous thickness and uniform color entire particle [18, 19, 52, 57]. Microscopes are even used for MPs less than 100 µm in size, with an accuracy rate of 63% for Particle sizes less than 50 µm, and 67% for Particle sizes between 50 and 100 µm [18, 19, 52, 57].

Fourier-transform infrared spectroscopy (FTIR): An efficient, simple, and non-destructive detection technique in which the type of the material (i.e., plastic or non-plastic) can be determined based on the generated characteristic spectrum and spectrum range by monitoring the specific vibration of the material structure [18, 19, 52, 57]. There are two operation modes, Transmission mode, and Reflection mode which only thick samples comprising regular shapes of a particular thickness of MP particles can be studied [18, 19, 52, 57].

Table 5 Methodologies used for the characterization of MPs [40]

Methodology	Advantages	Disadvantages
Fourier transform infrared coupled to microscopy (μ -FTIR)	Easy to use; Many particles can be analyzed simultaneously; Automatization available; Short time of analysis for single particles; Evaluation of size and shape; Detecting the intensity of oxidation; Report particles with shape and size information; Transmission and reflection mode; Nondestructive Less expensive than Raman and thermoanalytical techniques; Evaluation	Difficulty in characterizing black particles; Long time of analysis to measure multiple particles; Measures huge areas without particles; Detectors have to be cooled with liquid nitrogen; The analysis requires expert personnel; Huge data sets (several GB per filter); No total mass determination Expensive
Raman spectroscopy	Evaluation of size and shape; Many particles can be analyzed simultaneously; Automatization available; Detecting the intensity of oxidation Staining possible; Thermoelectrically cooled (TEC) detectors obviate the necessity for liquid nitrogen cooling; Report particles with shape and size information; Nondestructive; Filter contributions can be subtracted out; It is possible to detect additives, pigments, and plasticizers	More time-consuming measurements concerning FTIR-spectroscopy; The analysis requires expert personnel; Interference of biological and inorganic contaminants; No total mass determination; Expensive
Pyrolysis–Gas Chromatography–Mass spectrometry (Py–GC–MS)	The more holistic approach to characterize, in a single analysis, additives, and plasticizers, in addition to the polymer category; Powerful for mass determination	No particle number information; No evaluation of size and shape; Particles can be analyzed singularly; About 40 min of analysis for each particle determination; The analysis requires expert personnel; Destructive; Expensive
Thermal Extraction–Desorption–Gas Chromatography–Mass Spectrometry (TED–GC–MS)	The more holistic approach to characterize, in a single analysis, additives, and plasticizers, in addition to the polymer category; Powerful for mass determination	No particle number information; No evaluation of size and shape; Particles can be analyzed singularly; About 40 min of analysis for each particle determination; The analysis requires expert personnel; Destructive; Expensive

Attenuated Total Reflectance (ATR), ATR-FTIR: It is widely employed in the identification of microplastics since it can offer a strong signal-to-noise ratio and has an abundance of literature spectra. However, contact and pressure samples can destroy fragile microplastics, shifting samples from filters to rigid supports can result in missing or damaged samples, and imaging takes a long time and effort to discover MP particles acceptable for analytical work [18, 19, 52, 57].

Micro-FTIR: FTIR must be used in conjunction with an optical microscope in reflection or transmission mode for smaller particles with a typical size of up to 10 μm . Refraction flaws in irregularly formed microplastics will produce inexplicable spectra. Otherwise, these drawbacks are tough to overcome, and due to reflection faults induced by light scattering, the signal is disrupted/distorted [18, 19, 52, 57, 111].

FPA-FTIR: focal plane array (FPA)-based imaging: This method uses an infrared map to detect microplastics by scanning the surface of filters that contain microplastics. The use of a focal plane array (FPA)-based detection has improved FTIR imaging. This technique is unaffected by thickness and is unaffected by filter membranes or contaminants, making it an appropriate model for detecting microplastics [18, 19, 52, 57, 112–115]. Plastics smaller than 20 μm can be detected and identified using FPA imaging, with 5–10 μm being a more acceptable limit. In Reflection mode, it has a spatial resolution of 5.5 μm and 1 μm in ATR mode. Before performing an FPA analysis, the material must first be purified and then concentrated in a filter [18, 19, 52, 57, 112–115].

Raman spectroscopy: Thanks to the possibility of analyzing wet samples, the high spatial resolution which is device resolution down to 500 nm, and Confocal microscopy resolution can easily be 1 μm , Raman spectroscopies are widely used in microplastic studies in freshwater sediment [18, 19, 52, 57, 116].

Mass spectrometry analysis/pyrolysis gas chromatography-mass spectrometry (pyro-GC/MS)/TGA-MS: Thermoanalytical methods for the microplastic analysis of freshwater sediment do not require sample preparation, but they are ineffective for high impurity concentration mixes. They can also identify the type of chemical composition and the concentration of plastic-type, but not the number or shape of particles [18, 19, 52, 57, 117–120]. Samples smaller than 500 μm are also difficult to handle because they cannot be placed in a test tube [18, 19, 52, 57, 117–120]. Methods for mass analysis of some classes

of polymers based on preventive depolymerization and quantitative analysis of the resulting monomers have been proposed, specifically to overcome the dimensional limitations associated with analytical techniques, particularly spectroscopic ones, that are more widely used for the analysis of individual microplastics [18, 19, 52, 57, 117–120].

2.4.2 MPs characterization

Microplastic particles could be characterized by their size fractions and morphology and physical characterization of particles (referring to the origin or source, type, shape, color, and/or degradation stage of the particles).

Source and type of microplastics: Plastics are synthetic polymers manufactured from a wide range of chemicals with various properties. The most essential criterion for characterizing microplastic contamination is its chemical composition [65]. Plastic pellets were primarily obtained from plastic-processing plants near study sites, however weathering and biofouling may cause some features of plastic pellets, such as buoyancy and density, to change throughout their journey at sea, so the fact that many pelagic microplastics have different specific densities than main polymers demonstrate this [18, 19, 52, 57]. Microplastics with a high specific density (negative buoyancy) sink quickly in the open ocean, and so are absent from Neuston samples [18, 19, 52, 57]. Plastic consumer product fragments come in a variety of sorts and origins. Fishing nets, line fibers (polypropylene strands), thin plastic films, industrial raw material (e.g., from the ship-breaking industry), pellets, or polymer fragments of Oxo-biodegradable plastic could be the source of these particles [18, 19, 52, 57]. Microplastic polymers have been identified using a variety of approaches, including (FT-IR) and Near-infrared spectrometers can be used to compare the spectra of unknown tiny plastic pieces to those of recognized polymers [18, 19, 52, 57]; The crystalline structure of the polymer comprising small plastic fragments can be determined using Raman spectroscopy [18, 19, 52, 57]; Characteristic smoke during combustion and solvent assays are used to determine the polymers that microplastics are formed of [18, 19, 52, 57]; Based on density-based and color-based identification, synthetic polymers can also be identified using the specific density of the particles and, to a lesser extent, other features such as color [18, 19, 52, 57]. The plastic piece is placed in distilled water and titrated with ethanol or concentrated calcium or strontium chloride solutions until it is neutrally buoyant [18, 19, 52, 57]. Because those properties have been described for

virgin pellets, rapid and cost-effective polymer identification of plastic pellets is possible with this method [18, 19, 52, 57]; (ATR) FT-IR spectroscopy can identify irregularly shaped microplastics that cannot be recognized by FT-IR spectroscopy, however, the main disadvantage is the expensive cost of this equipment. Plastic fragments, pellets, filaments, plastic films, foamed plastic, granules, and Styrofoam are the MPs classification based on their types [18, 19, 52, 57].

Shape and erosion of microplastics: In the case of shape, originally the MPs were classified as irregular, elongated, degraded, rough, and broken edges particles [18, 19, 52, 57]. Based on their form for pellets they are classified as cylindrical, disk, flat, ovoid, and spheroids; and for fragments, they are considered rounded, sub-rounded, sub-angular angular particles [18, 19, 52, 57]. UV light, photo-degradation, and biodegradation Chemical weathering, or Physical forces (wave action, wind, sand-blasting), Time spent in the environment, Strain, and Fatigue, the original form of primary MP, could be effective on cracking, degradation, and erosion of the particle surface, as well as producing a wide variety of particle shapes, including pellet/spherule (spherule and microbeads used in cosmetic products and sandblasting media, as well as in air-blasting agents or industrial cleaner); fragment/sheet; foam (from Styrofoam damage), fiber(from washing)/line; and film (from plastic bags and packaging materials) [18, 19, 52, 57]. Moreover, in case of erosion, the MPs classification would be a fresh, un-weathered, incipient alteration, and level of crazing (conchoidal fractures), weathered, grooves, irregular surface, jagged fragments, linear fractures, subparallel ridges, and very degraded [18, 19, 52, 57]. According to the observations, smaller particles were consistently more circular, while larger particles displayed more elongated forms and/or uneven surfaces. Angular and sub-angular particles showed conchoidal fractures, while rounded particles had linear fractures and adhering particles, according to scanning electron microscopy [18, 19, 52, 57]. Continuous particle-particle collision may be the cause of numerous surface scratches on primarily degraded angular plastic pieces (<1 cm²) [18, 19, 52, 57]. Eroded or weathered plastic pellets are pellets that have been weathered to some extent [18, 19, 52, 57]. Although plastics undergo numerous physical and chemical changes as they age, pollutant sorption on plastic pellets increases with surface area because of weathering, which increases the sorbate's effective diffusivity. The surface roughness of fragments and pellets (for example, grooves, fissures,

attached particles, and flakes) shows mechanical wear and chemical weathering to form microplastics [18, 19, 52, 57].

Color of microplastics: Plastics are found in different colors like transparent, crystalline, white, clear-white-cream, red, orange, blue, opaque, black, gray, brown, green, pink, tan, yellow, and pigmentation. Colors were employed to identify the chemical components of the most frequent pellets as a first step and can facilitate separation, as well as eye-catching colors, making the particles more distinguishable among the sample [18, 19, 52, 57]. Aquatic species misinterpret colored microplastic particles for food easily. Because the discoloration process (yellowing) is indicative of a longer exposure time to seawater, which increases the chances of the polymers becoming oxidized, color has also been used as an index of photo-degradation and residence time at the sea surface, as well as the degree of tarring or weathering [18, 19, 52, 57].

Size fractions of microplastics: Microplastics' particle size has a direct impact on their migration in the aquatic environment and whether they can be consumed by organisms, which is connected to biosafety [23, 65, 99]. Microplastic particles less than 1 mm are more frequent in freshwater sediments, with abundances decreasing as particle size increases, according to studies. The National Oceanic and Atmospheric Administration (NOAA) proposed in 2008 that microplastics be defined as particles smaller than 5 mm [52]. Pellet sizes determine size restrictions. The diameter of plastic preproduction pellets ranges from 1 to 5 mm, with an average diameter of 3.5 mm [14, 18, 19, 52, 57]. Microplastics come in a variety of sizes that are similar to those seen in benthos and plankton ecosystems (micro-, meso-, and macrofauna for benthos, and micro- and macroplankton). The same may be said for geological sediment types (silt and sand), which are mobilized in the same way that microplastics are [14, 18, 19, 52, 57]. Particles were 0.25 to 5 mm in diameter, and many were also >4.8 mm sea surface samples. Particles larger than 500 micrometers are kept in ordinary sieves and sorted using a dissecting microscope. Particles smaller than 500 microns were typically only obtained through density separation and filtration investigations, while particles smaller than 2 microns were unlikely to be collected representatively [14, 18, 19, 52, 57]. Microplastics have no minimum size requirements. In sediment samples, the smallest size documented was 1 μm in diameter and 20 μm in length. For sediment samples, most investigations reported values above 500 μm, and for seawater samples, values above 300 μm [14, 18, 19, 52, 57].

2.5 Quality assurance and quality control

To avoid contamination, take precautions during the *sample collection* (secondary contamination from synthetic fibers in clothing or polymer particles in the air, ambient synthetic fibers), *preparation* (leakage from the filter, adhesion MPs in transfer and filtration devices), and *analysis processes* [18, 19, 52, 57, 104, 108, 121–126]. Several prior studies have provided many important ideas for improving QA/QC procedures in the field as well as in laboratories for microplastic analysis, which comprise three broad areas. The establishment of effective field and laboratory methods aimed at avoiding procedural MP contamination is covered in **the first part** such as, when possible, polymer-manufactured equipment was eliminated, and metal or glass alternatives were used. Before usage, inspect chemicals and filtration solutions, sanitizing tools and working areas. Regular checks of washing solutions are required to ensure that they are free of polymers. Decontaminated water and ethanol were used to sanitize surfaces and equipment before being analyzed. Before usage, water and other liquid ingredients were filtered. When possible, samples and instruments were wrapped with aluminum foil. Reducing the laboratory employees. When dealing with MP samples, keep all windows closed. Throughout the sample, treatment, and analysis, appropriate clothes such as cotton jackets, headwear, masks, and nitrile gloves are worn. Before usage, clean laboratory clothing using compressed air or filtered water/alcohol solutions. If feasible, work in a clean room environment following its protocols [18, 19, 52, 57, 104, 108, 121–126]. The quantification of background contamination using field and procedural blanks is **the second section** that must be improved. During the experiment, the

blank is measured, and the environmental blank is subtracted from the result to account for background contamination. The measured concentration of the sample is used to calculate the average value of the repeated samples. Various microscopic methods were used to investigate the particles on the filters and in the sand, and the results were normalized to the final MP values [18, 19, 52, 57, 104, 108, 121–126]. Method validation utilizing positive controls is **the third section**, which is important for further strengthening QA/QC methods. Each step must be validated before proceeding. Positive controls, such as MP spiked blanks, are valuable in the field and laboratories for evaluating existing procedures. As reference material, a diverse range of polymers in varied shapes, colors, and sizes should be examined [18, 19, 52, 57, 104, 108, 121–126].

3 Conclusions

As a result, due to the microplastic cytotoxicity, acute reactions, and adverse immunological responses, they constitute a major health concern to marine creatures and humans. Aquaculture, agriculture, fisheries, transportation, industrial sectors, power generation, tourism, and local governments are all disrupted, resulting in substantial financial losses. Microplastics can be reduced by identifying major sources and concentrations of environmental plastic pollution using various methodologies, as well as educating the public, resulting in a reduction in microplastic emissions into the environment. Furthermore, exploiting the potential of microorganisms, particularly those marine sources that can digest plastics, could provide a more effective and environmentally acceptable approach to reducing microplastic contamination.

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