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Global insights into microplastic contamination in marine life: detection methods and current status

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Microplastics (MPs) pollution has become a pressing environmental issue, significantly impacting marine ecosystems and food safety. These synthetic particles, defined as plastic fragments smaller than 5 mm, originate from various sources and have infiltrated marine habitats worldwide, from surface waters to deep-sea sediments. This review provides a comprehensive analysis of MPs contamination in marine organisms, highlighting global case studies and detection methodologies. MPs are ingested by marine life through direct uptake or trophic transfer, causing adverse biological effects, including growth inhibition, reproductive impairments, and bioaccumulation of toxic substances. Humans are exposed to MPs primarily through seafood consumption, raising concerns about potential health risks. We examine various detection techniques, including visual identification, spectroscopic methods (FTIR, Raman spectroscopy), and thermal analytical approaches (Py-GC/MS, TGA), evaluating their advantages and limitations. Despite significant research progress, challenges remain, particularly in detecting nanoplastics, standardizing methodologies, and understanding the long-term ecological and human health implications. Case studies from different regions demonstrate varying contamination levels, influenced by local environmental conditions, industrial activities, and waste management practices. This review emphasizes the need for improved monitoring, regulatory frameworks, and mitigation strategies to address the pervasive threat of MPs pollution. Future research should focus on refining detection technologies, assessing human health impacts, and implementing policy measures to reduce plastic emissions into marine environments.

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Introduction

Plastic pollution has emerged as a critical environmental threat, particularly in marine ecosystems, due to the widespread use and persistence of synthetic polymers.[1] Originating from products first developed in the late 19th century[2] and mass-produced post-World War II,[3] plastics are now ubiquitous. Their durability, once an advantage, has resulted in long-term environmental contamination.[4] Over time, larger plastic debris degrades into microplastics (MPs)—plastic fragments smaller than 5 mm (ref. 5)—and even nanoplastics at the sub-micron level.[6] These particles have been detected across all environmental compartments, including surface waters,[7] sediments,[8] deep-sea habitats,[9] soils,[10] and the atmosphere. According to Jambeck *et al.*, an estimated 8 million metric tons of plastic waste enter the oceans annually, contributing to the generation of trillions of microplastic particles worldwide. Global modeling studies suggest that the oceans currently contain between 15 and 51 trillion microplastic particles, weighing approximately 93 000 to 236 000 metric tons.[11,12] To visualize these data, Fig. 1 presents a summary of global plastic waste inputs and modeled marine distribution of MPs. Moreover, a UNEP 2021 report highlights that plastic accounts for up to 85% of total marine litter, underscoring its pervasive presence. Their infiltration into the food chain is of growing concern, with MPs found in seafood species consumed by humans,[13] drinking water,[14] and various food products such as table salt and honey,[15] raising alarms about food safety and potential human exposure.[14]

Marine organisms encounter MPs directly through their environment. Filter feeders (*e.g.*, mussels and oysters) ingest suspended MPs,[16] while predatory species acquire them *via* trophic transfer.[1] Numerous studies report that MPs ingestion negatively affects marine life, manifesting as reduced feeding, growth inhibition, reproductive impairment, behavioral

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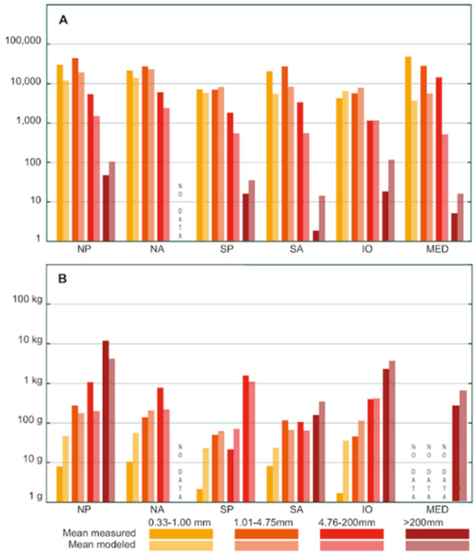
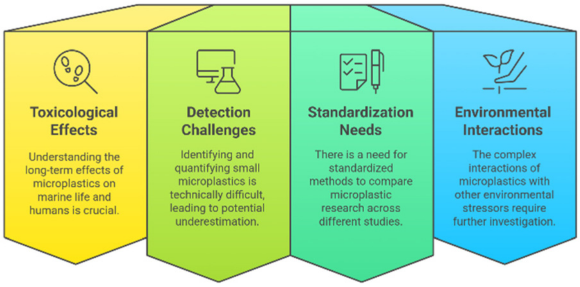


Fig. 1 Comparison of mean and modeled densities. Comparison of dataand model predictions for count density (A – pieces km-2) and weight density (B – weight km-2) for four size classes from six ocean regions: North Pacifc (NP), North Atlantic (NA), South Pacific (SP), South Atlantic (SA), Indian Ocean (IO), and Mediterranean Sea (MED).[12]

changes, and increased mortality.[13] These biological effects pose significant risks to marine biodiversity[17] and, by extension, to seafood safety.[18] The health effects of human exposure to MPs are still uncertain. However, studies indicate they may carry harmful chemicals or pathogens and can reach vital organs like the liver and brain.[14] This highlights an urgent need to understand the toxicological risks posed by MPs to both marine organisms and human health.[19]

Research on MPs contamination in marine life has expanded rapidly in the past decade,[20] with scientists reporting MPs in hundreds of marine species across all trophic levels. However, critical knowledge gaps remain (Fig. 2). One major gap is understanding the long-term toxicological effects of chronic MPs exposure on marine organisms and humans. While ecotoxicological studies indicate harm to marine life, the dose thresholds at which MPs pose a serious risk to wildlife or human consumers have not been established. In fact, regulatory agencies have yet to determine a safety threshold for MPs in food due to insufficient data. Another gap lies in the analytical methods: detecting and quantifying the smallest MPs (especially nanoplastics <1 μm) is technically challenging, leading to possible underestimation of true contamination levels.[21] Standardized methods for sampling and analysis are needed to compare results across studies and regions. MPs interact with other environmental stressors such as climate change and pollution. For example, they often carry pollutants or serve as surfaces for microbial colonization, increasing their environmental impact.[22] These uncertainties underscore the need for comprehensive research.


Toxicological Effects
Understanding the long-term effects of microplastics on marine life and humans is crucial.

Detection Challenges
Identifying and quantifying small microplastics is technically difficult, leading to potential underestimation.

Standardization Needs
There is a need for standardized methods to compare microplastic research across different studies.

Environmental Interactions
The complex interactions of microplastics with other environmental stressors require further investigation.

Fig. 2 MPs research gaps.

In the following sections, we provide a global overview of MPs contamination in marine life, emphasizing detection methods and current status. We first review the detection and analytical methods used to identify and quantify MPs in environmental and biological samples, comparing their strengths and limitations. Next, we survey global case studies, highlighting regional patterns of contamination in seafood and marine fauna and discussing potential human exposure risks. We then delve into the mechanisms and underlying factors that influence MPs impacts, including biological uptake pathways, the role of biofilms, and interactions with other stressors. Finally, we conclude with a synthesis of key findings and discuss perspectives, including policy recommendations and future research directions needed to address this emerging pollutant. By consolidating global insights, we aim to clarify the current status of MPs pollution in marine life and identify strategies to safeguard marine ecosystems and food safety.

Sampling, isolation, and detection techniques

Accurately detecting and quantifying MPs in marine environments and organisms is fundamental for understanding contamination levels. However, MPs present unique analytical challenges due to their small size, diverse shapes and polymer types, and tendency to accumulate in complex matrices (water, sediment, or biological tissue). Over the years, researchers have developed a suite of methods for sampling and analyzing MPs. These methods can be broadly grouped into: (1) sampling and isolation techniques, (2) visual and microscopic identification, (3) spectroscopic identification, (4) thermal analytical techniques, and (5) emerging or combination methods. Each approach has distinct advantages and drawbacks, and often multiple methods are used together to improve reliability.

However, as the complexity of environmental matrices increases—such as highly organic-rich sediments, biota with strong pigment interferences, or the presence of nanoplastics —conventional methods may become insufficient. In such cases, advanced detection technologies have proven essential. Techniques like thermal extraction desorption-gas chromatography–mass spectrometry (TED-GC/MS) enable polymer-specific identification with minimal sample preparation, while emerging bioresponsive sensors and aptamer-based platforms offer selective and rapid detection in biological tissues. Moreover, machine learning-enhanced image recognition systems now facilitate automated classification of MPs from large datasets with reduced subjectivity. These technologies address key challenges in throughput, resolution, and polymer specificity, and are increasingly used in tandem with traditional methods to ensure comprehensive and accurate MPs characterization, especially in samples with high background interference.

To facilitate an at-a-glance comparison of key methods discussed, we have included Fig. 3, which presents a decision matrix that evaluates commonly used MPs detection techniques across a range of practical criteria. This matrix is intended to assist researchers in selecting appropriate techniques based on study goals (*e.g.*, polymer identification *vs.* particle quantification), sample type (*e.g.*, sediment, biota, water), and resource availability.

Sampling and isolation techniques

The first step in MPs analysis is obtaining representative samples from the environment or biota. Sampling methods must be carefully chosen because they strongly influence the results.[23] In aquatic environments, surface water MPs are commonly collected using nets such as manta trawls or neuston nets that skim the water surface. These nets (with mesh sizes typically around 300 µm) retain floating plastic pieces, especially larger MPs (>0.3 mm), over a towed distance. For subsurface or deeper waters, specialized pumps or plankton nets can be used at various depths. To collect MPs from sediments or beaches, researchers use tools like corers or scoops. The samples are then sieved and processed for further analysis. Biotic samples (marine organisms destined for MPs analysis) require careful handling to avoid contamination from airborne plastics or equipment. Laboratory protocols typically involve dissecting organisms (*e.g.*, fish gastrointestinal tracts, bivalve tissues) and then isolating MPs by digesting organic material. Chemical digestion uses reagents such as hydrogen peroxide or strong alkalis/acids (*e.g.*, 10% KOH or enzymatic digestion) to dissolve soft tissues, leaving behind MPs particles.[24] The residue can then be filtered so that MPs remain on a filter membrane for further analysis. Throughout sampling and isolation, strict quality assurance/quality control (QA/QC) measures are recommended, such as procedural blanks and using non-plastic tools, to prevent introduction of extraneous plastic fibers. Fig. 4 shows the scheme of MPs sampling and analysis process. These steps ensure that detected MPs indeed originate from the sample and not the laboratory environment.

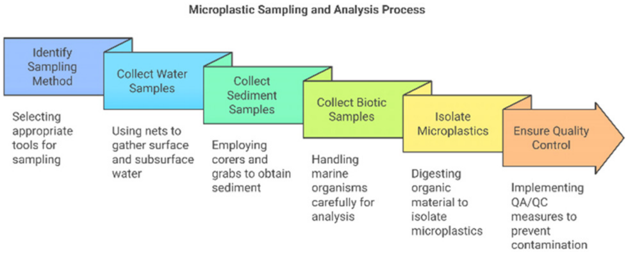


Fig. 4 Scheme of MPs sampling and analysis process.

Visual and microscopic identification

Once a sample is prepared (*e.g.*, filtered onto a membrane or sorted from sediment), visual inspection is often the first approach to identify suspected MPs. Under a stereomicroscope (Fig. 5) or even the naked eye, MPs can be recognized by their physical characteristics: they often have no cellular or organic structures, may have unnaturally bright colors, and exhibit textures (films, fragments, fibers, beads) distinct from natural materials. Simple physical tests (prodding with a needle to test flexibility, for instance) can help distinguish plastic fragments from organic debris. Light microscopy is widely used to examine smaller MPs (tens of micrometers and above).[25]

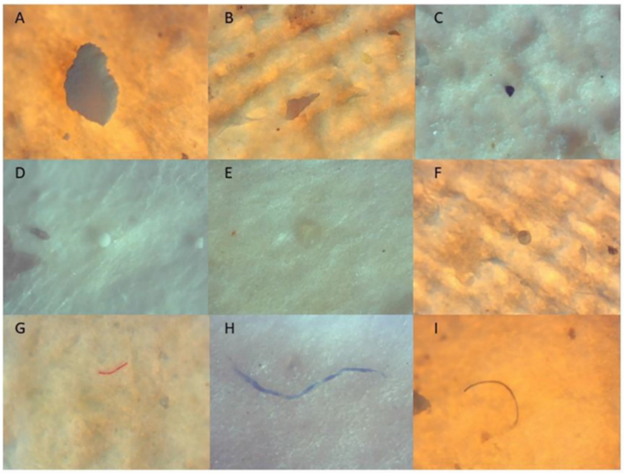
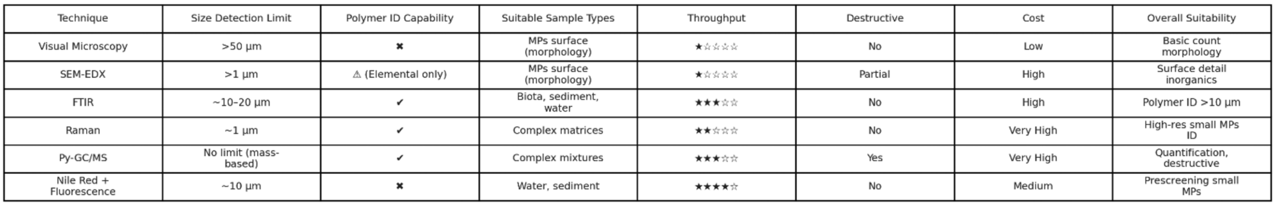


Fig. 5 Examples of MPs found in individuals of *Anomalocardia flexuosa* using a stereomicroscope[29].



Technique	Size Detection Limit	Polymer ID Capability	Suitable Sample Types	Throughput	Destructive	Cost	Overall Suitability
Visual Microscopy	>50 µm	✖	MPs surface (morphology)	★☆☆☆☆	No	Low	Basic count morphology
SEM-EDX	>1 µm	⚠ (Elemental only)	MPs surface (morphology)	★☆☆☆☆	Partial	High	Surface detail inorganics
FTIR	~10–20 µm	✔	Biota, sediment, water	★★★☆☆	No	High	Polymer ID >10 µm
Raman	~1 µm	✔	Complex matrices	★★☆☆☆	No	Very High	High-res small MPs ID
Py-GC/MS	No limit (mass-based)	✔	Complex mixtures	★★★☆☆	Yes	Very High	Quantification, destructive
Nile Red + Fluorescence	~10 µm	✖	Water, sediment	★★★★☆	No	Medium	Prescreening small MPs

Fig. 3 Decision matrix for selecting microplastic detection techniques based on practical considerations.

Under a microscope, researchers categorize MPs by shape (*e.g.*, fiber, fragment, bead, film) and measure their size. This method enables direct counting and sizing of particles. Microscopic analysis is intuitive and does not require highly specialized instruments, but it has notable limitations. The advantages include relatively simple sample preparation and the ability to directly see the particles, providing immediate confirmation of MPs presence.[26] However, microscopy is time-consuming and labor-intensive, often requiring an analyst to manually pick out and identify each particle.[27] This can lead to subjectivity – results may vary between researchers depending on their experience and judgment, especially for tiny, transparent particles that are hard to differentiate from organic matter. Consequently, purely visual methods tend to have lower accuracy and reproducibility compared to spectroscopic methods. For example, distinguishing a small clear plastic fragment from a sand grain or shell fragment by eye can be error-prone.[28] Additionally, visual methods alone cannot confirm the polymer type of a suspect particle. Despite these drawbacks, microscopy remains a foundational step in MPs research and is often coupled with other techniques. Many studies first isolate and count MPs under a microscope, then select a subset for chemical identification by spectroscopy to confirm the particles are plastic and determine their composition.

To enhance visual detection, especially for MPs that are too small or transparent to easily see, fluorescent staining techniques have been developed. One common approach uses dyes like Nile Red, which selectively bind to hydrophobic plastic polymers and fluoresce under specific wavelengths of light. By staining a sample with Nile Red and viewing it under a fluorescence microscope or blue-light source, MPs light up and can be more readily distinguished from non-plastic debris. This method has been reported to recover a high percentage of MPs in environmental samples while being faster than exhaustive manual sorting.[30] Its drawback is that some organic materials may also take up the dye, and thus staining is usually combined with spectral confirmation or careful controls to avoid false positives.[31] Nonetheless, fluorescence tagging is an emerging, cost-effective way to screen samples for MPs, especially the smaller size fractions that might otherwise be overlooked.

When greater detail on morphology is needed, or to visualize MPs at the nanoscale, electron microscopy comes into play. Scanning Electron Microscopy (SEM) can produce high-magnification, high-resolution images of MPs surfaces. Under SEM, researchers can observe fine details like surface weathering (cracks, pits) which indicate aging, or biofouling layers on the plastic. SEM is often paired with Energy-Dispersive X-ray spectroscopy (EDX/EDS), which provides elemental composition analysis of the sample. SEM-EDX can confirm a particle is synthetic by showing elements like chlorine (for PVC) or high carbon signals. It can also detect inorganic fillers like titanium dioxide.[32] Fig. 6 shows the SEM images and corresponding EDX spectra of acrylonitrile butadiene styrene (ABS), polypropylene (PP), and polyethylene (PE). Because most polymers are non-conductive, SEM-EDX analysis requires coating the sample with a conductive layer like gold or carbon to prevent electron beam charging. This extra preparation is time-intensive and means the sample is no longer in its original state (and cannot be reused for other analyses). SEM-EDX identifies elements rather than molecular structures. For instance, it can show carbon and oxygen in a particle but cannot tell whether it's PE or natural material.[33] It also cannot discern plastic color (important if color is used as a diagnostic feature). Although SEM-EDX offers high-resolution imaging and elemental analysis, it is typically reserved for special cases due to its cost, sample preparation time, and destructive nature.

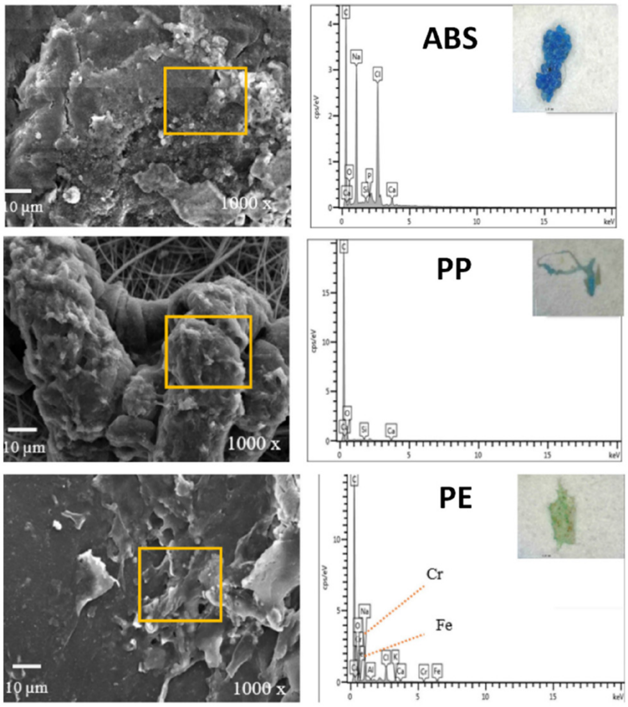


Fig. 6 SEM images and corresponding EDX spectra of ABS, PP, and PE.[34]

Spectroscopic identification (FTIR and Raman)

To unambiguously identify MPs and determine their polymer composition, spectroscopic techniques are widely employed. The two most common are Fourier-transform infrared (FTIR) spectroscopy and Raman spectroscopy. Fig. 7 shows examples

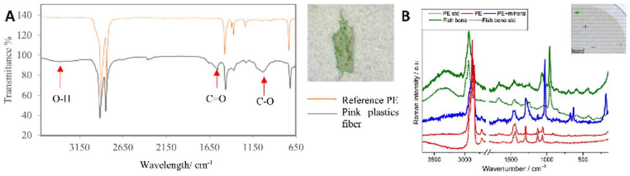


Fig. 7 (A) FTIR spectra recorded from reference PE and pink plastics fiber.[34] (B) Raman spectra of reference PE, fish bone standard and MPs sample.[35]

of FTIR and Raman spectra recorded for MPs. Both are forms of vibrational spectroscopy that detect the unique molecular vibrations of materials, effectively providing a "fingerprint" for different polymers.

FTIR shines infrared light onto a sample and measures the wavelengths that are absorbed, which correspond to specific bond vibrations in the material. Each type of plastic (PE, PP, polystyrene, *etc.*) has characteristic absorption peaks in the IR spectrum. By comparing the sample's IR spectrum to reference spectra in libraries, researchers can identify the polymer. FTIR can be used in different modes. ATR-FTIR involves pressing particles onto a crystal so that infrared light interacts with the surface, producing a spectrum that can help identify the polymer. ATR-FTIR is generally effective for particles ≥20 μm, as it requires sufficient contact between the particle surface and the ATR crystal. In practical applications, particles smaller than 10–20 μm often yield weak or distorted spectra due to poor contact and insufficient penetration depth, which limits the accuracy of identification.[36] In contrast, micro-FTIR (also known as FTIR imaging), particularly when equipped with focal plane array (FPA) detectors, can routinely detect and identify MPs down to ∼10 μm in real samples, and in some optimized settings, even approach detection near 5–6 μm. For instance, Prata *et al.*[37] reported successful characterization of MPs as small as 10 μm using micro-FTIR with transmission mode and a high-resolution detector. This makes micro-FTIR more suitable for detailed analysis of smaller MPs and for processing a large number of particles on a single filter. However, it requires higher instrumentation cost, longer processing times, and extensive sample preparation to reduce background interference. In contrast, ATR-FTIR is more accessible and faster for identifying larger, isolated particles but is less suitable for high-throughput or fine-resolution imaging applications.

The advantages of FTIR are clear: it provides specific chemical identification, is non-destructive (the particles remain intact after analysis), and can process multiple particles at once with imaging systems. It also can sometimes give semi-quantitative data on additives if they have distinctive IR peaks (for example, certain flame retardants or plasticizers might be detectable). However, FTIR has drawbacks. It can be time-intensive for samples with thousands of particles since high-resolution scans are needed for each particle or area of interest.[38] The equipment (especially FTIR microscopes with imaging capabilities) is expensive and requires trained operators. Small or irregularly shaped MPs can produce weaker or distorted spectra – for instance, ATR-FTIR can suffer if a particle doesn't make perfect contact with the crystal, and very small or thin particles may produce weak signals in transmission mode.[39] In practice, samples often need to be cleaned before FTIR (to remove biofilms or inorganic residue) because attached organic matter or sand grains can interfere with the spectrum. Another challenge is the possibility of misidentifying materials: spectra of some substances can mimic plastics.[40] As a cautionary example, certain surfactants or biogenic materials like chitin have IR peaks that could be confused with polymers if the reference libraries are incomplete. Therefore, expanding spectral libraries (including spectra of common impurities or biogenic materials) and analyst expertise are important to minimize false positives.

Raman spectroscopy is complementary to FTIR. Instead of absorption, Raman measures light scattering: a laser is shone on the particle and the spectrum of scattered light (with shifts in wavelength corresponding to molecular vibrations) is recorded. Like FTIR, every polymer yields a characteristic Raman spectrum that can be matched to a library for identification. Raman microscopy focuses a laser to a very small spot (on the order of 1 μm or less), giving it an edge in detecting very tiny particles – even down to the micron or sub-micron scale, which pushes into the nanoplastic range that FTIR might miss. Raman is also a non-contact method, so it preserves the sample and avoids potential mechanical damage. This is useful if one wants to perform additional analyses on the same particle after Raman, or simply to keep it as evidence.[41] However, Raman spectroscopy also has its limitations. One issue is that many real-world MPs contain pigments or dyes and other additives. These additives sometimes dominate the Raman signal or cause fluorescence that masks the Raman peaks, making it hard to identify the base polymer. For instance, a blue PE fragment might have a Raman spectrum where the signal from the blue dye obscures the characteristic peaks of PE.[42] Additionally, Raman, like FTIR, relies on good reference spectra and can be confounded if two materials have similar spectral features. An example cited in the literature is confusion between PE and certain long-chain fatty acid compounds (like stearates) – a reminder that even Raman can produce false positives if a non-plastic particle has a coincidentally similar spectrum.[43] Another practical drawback is sample heating or degradation: the laser in Raman can be intense, and if focused on a small dark-colored plastic, it might locally heat or burn the particle, altering it or reducing quality of data (this often can be mitigated by using lower laser power or shorter exposure). Finally, the instrumentation is high-end and requires specialized expertise to interpret spectra, similar to FTIR in cost and skill.

In summary, FTIR and Raman spectroscopy are indispensable for confident identification of MPs, each with trade-offs. FTIR is generally better for analyzing many or larger particles, while Raman provides higher resolution for tiny ones and serves as a useful complementary method. FTIR and Raman spectroscopy provide chemical verification of MPs. This is essential because visual identification alone can be misleading or inaccurate. Despite the time and cost, these spectroscopic methods greatly improve the rigor of MPs detection by providing polymer-specific information.

Thermal analytical techniques

While microscopy and spectroscopy focus on individual particles, thermal analysis methods take a different approach: they analyze bulk samples by heating them and detecting thermal decomposition products. These methods are particularly useful for quantifying total MPs content in a sample (by mass) and identifying polymer types based on thermal behavior or evolved gases. Key thermal techniques include Thermogravimetric Analysis (TGA), Differential Scanning Calorimetry (DSC), and Pyrolysis Gas Chromatography–Mass Spectrometry (Py-GC-MS), often used in combination.

In TGA, the sample (*e.g.*, a filtered residue suspected of containing MPs) is heated at a controlled rate, and the weight loss is recorded continuously. Different polymers decompose or combust at characteristic temperature ranges, so a multi-step weight loss curve can indicate which polymers were present. For example, a distinct weight loss around 400 °C might suggest the decomposition of PVC or PE, whereas a loss at 300 °C could indicate another polymer. DSC can similarly detect thermal transitions like melting points. However, in environmental samples with mixed polymers, overlapping decomposition ranges can make it hard to deconvolute signals. Studies have found TGA/DSC alone might clearly identify some common plastics like PE and PP because their thermal decomposition stands out,[44] but could miss or confuse others (polyamide, polyester, *etc.*) if their thermal signals overlap or are masked.[45] Purely thermal analysis without additional techniques has limited resolution when multiple polymer types are present.

Pyrolysis-GC/MS adds a powerful dimension to thermal analysis. In this method, the sample is heated (pyrolyzed) to a very high temperature (often ∼600–700 °C) in an oxygen-free environment, causing all polymers to break apart into smaller, volatile compounds. These gaseous decomposition products are then separated by gas chromatography and identified by mass spectrometry. The resulting chromatogram (pyrogram) is essentially a chemical fingerprint of the original material. Different polymers yield different signature compounds when pyrolyzed. For example, polystyrene (PS) yields characteristic styrene fragments;[46] PE yields a series of aliphatic hydrocarbons,[47] *etc*. By matching the pattern of pyrolysis products to known standards, one can infer which polymers were in the sample. Py-GC/MS is highly sensitive and only a very small amount of sample (on the order of a few milligrams or even less) is needed to detect polymers. It has been used to confirm the presence of various plastics in environmental matrices—such as sediments, biota, and wastewater—especially when visual or spectroscopic methods are limited.[48] However, a notable drawback is the potential for polymer misidentification due to overlapping pyrolysis products. For instance, polyester and polyamide may produce similar decomposition compounds that can complicate accurate polymer differentiation unless compound-specific markers are well-characterized.[49] Moreover, the high operational cost and requirement for advanced instrumentation and expertise limit the routine applicability of Py-GC/MS in standard monitoring programs. Most critically, thermal methods are inherently destructive; they completely degrade the sample, thus precluding further analysis of particle morphology, size distribution, or reusability for cross-validation. As such, while valuable for quantifying total polymer content and confirming polymer classes in complex mixtures, thermal techniques are best employed as complementary tools rather than standalone solutions in MPs analysis pipelines. Py-GC/MS is effective in detecting polymers missed by visual or FTIR methods, including weathered or very small particles that still contribute to the chemical signature. Py-GC/MS quantifies MPs by mass. For example, it can determine the microgram amount of PE in a sample rather than just count particles. This is useful for risk assessment since the mass might correlate with effects.

However, Py-GC/MS and other thermal methods are destructive – the sample is entirely consumed by heating, meaning one cannot retrieve the actual particles afterward. As such, these methods do not provide information on the number of particles or their sizes and shapes, just the total quantity and types of polymers present. Another limitation is that some polymers may not have unique pyrolysis markers unless the Py-GC/MS method is carefully optimized. Researchers note that while PS and related plastics are readily identified, other polymers might need specific marker compounds to be identified with confidence.[50,51] If those markers are not known or if multiple polymers produce similar breakdown products, interpretation can be tricky. Additionally, traditional Py-GC/MS can only analyze a small sample size (a few mg), which might not be fully representative of an environmental sample. To overcome sample size limitations and to marry the strengths of spectroscopy and thermal analysis, hybrid techniques have been developed. One example is TGA-FTIR-GC/MS, where the off-gases from a TGA are continuously analyzed by FTIR and then captured for GC/MS analysis.[52] This setup allows real-time monitoring of decomposition (qualitative and quantitative) and a detailed breakdown of components, providing a very comprehensive analysis. Another advanced method is TED-GC/MS (thermal extraction-desorption GC/MS).[53] In TED-GC/MS, a relatively large sample can be heated in steps; the evolved gases from each step are trapped, then released into a GC/MS. This allows much larger sample masses to be analyzed. The advantage is improved detection of low concentrations of MPs and capturing even very small particles that collectively contribute to the mass. Using such combined methods, researchers have been able to identify multiple polymer types in one analysis and even quantify total MPs content by polymer class (PE, PP, PS, PVC, *etc.*). For instance, a study using TED-GC/MS showed it could detect MPs down to 5 μm in size that might evade FTIR detection, by analyzing the total outgassed products.[54]

Comparison and emerging techniques

In MPs research, each detection technique has its own strengths and limitations. Consequently, researchers often select or combine multiple methods depending on the study's objectives and the characteristics of the sample. Microscopic methods reveal particle shape and count. However, identifying polymers can be subjective without chemical confirmation methods. FTIR and Raman distinguish polymers *via* vibrational fingerprints. Yet, analyzing many or very small particles requires time and specific preparation. Thermal analysis gives total polymer mass in a sample. But it destroys particles, so size, shape, and count data are lost.

Because a single approach rarely captures all necessary information about MPs, researchers typically integrate methods. For instance, combining visual or microscopic counting with spectroscopic analyses not only yields abundance and size distribution data but also clarifies polymer identities.[55] Thermal techniques, in turn, can complement these results by revealing total MPs mass and confirming polymer classes in heterogeneous samples.[56] This multipronged strategy ensures a more holistic assessment of MPs contamination. Beyond these established methods, new technologies are emerging to overcome current bottlenecks. Fluorescent dyes like Nile Red stain MPs for easier counting. Machine learning tools can speed up and reduce bias in particle classification.[57] Nonetheless, detecting nanoplastics remains an ongoing challenge due to the limitations of spatial resolution in most current analytical approaches. Potential solutions include high-resolution techniques like AFM-IR (atomic force microscopy-based infrared spectroscopy)[58] or biosensors[59] with strong binding selectivity to polymer surfaces. Although these novel strategies are promising, many are still in the experimental stage and require further validation. To facilitate an at-a-glance comparison of key methods discussed, Table 1 summarizes their fundamental principles, typical detection ranges, primary advantages, and main drawbacks. Visual and microscopic methods show particle morphology. Spectroscopy verifies composition but is slower. Thermal analysis gives mass but not particle-level detail. Therefore, combining multiple methods is often essential to obtain reliable, comprehensive information on MPs contamination. Advances like high-resolution imaging, better dyes, and AI classification are improving MPs detection and quantification across environments. As these techniques continue to evolve, so too will the accuracy and comparability of MPs data gathered worldwide, ultimately informing stronger risk assessments and policy decisions.

Recent global advances in analytical techniques for microplastics detection

While conventional analytical methods (*e.g.*, FTIR, Raman spectroscopy, Py-GC/MS) have significantly contributed to our understanding of MPs contamination, recent years have seen a surge in technological innovations aimed at overcoming current limitations—especially in detecting smaller particles, improving throughput, and enhancing automation and accuracy.

One promising avenue involves machine learning (ML) and artificial intelligence (AI) integrated with microscopic imaging. Image recognition software powered by convolutional neural networks (CNNs) can now classify MPs by shape and size from high-resolution images with improved speed and reduced sub-

Table 1 Comparison of common MPs detection techniques

Method	Principle	Typical particle size range	Major advantages	Major limitations
Visual & microscopic approaches	Direct observation of particle morphology (optical microscopy, SEM-EDX); fluorescent dyes for improved contrast	Generally >20–50 μm (SEM can go smaller)	- Simple sample preparation - Enables direct assessment of shape and size	- Can be labor-intensive and subjective - Difficult to confirm polymer type (requires complementary methods) - Small or transparent particles can be missed
Spectroscopic methods (FTIR, Raman)	Identification of polymers by molecular vibrational "fingerprints" (infrared absorption or Raman scattering)	FTIR ∼10 μm; Raman ∼1 μm (or smaller)	- Confident polymer identification - Non-destructive, preserves particles - Raman excels at sub-micrometer resolution	- Instruments can be expensive and require specialized expertise - Analysis of large numbers of particles can be time-consuming - Pigments and fluorescence may obscure signals
Thermal analysis (Py-GC/MS, TGA, DSC)	Heating samples to decompose polymers; measuring weight loss or analyzing released pyrolysis products (gas-phase)	No strict lower size limit (mass-based)	- Can handle complex mixtures - Provides total MPs mass fraction - Py-GC/MS is highly sensitive, can detect multiple polymer types	- Destructive technique; cannot retrieve particle shape/size/number - Overlapping signals may complicate interpretation - Often requires extensive calibration and high operational costs
Emerging & hybrid methods	Includes fluorescent staining, automated image analysis, AFM-IR, biosensors, *etc.*	Method-dependent; target down to nano-scale	- Higher resolution or automated processing - Potential for nanoplastic detection - Faster, more objective identification	- Many are still in development, requiring validation - May involve expensive equipment and specialized protocols - Accuracy and reproducibility need further demonstration

jectivity, particularly in large environmental datasets.[60] For instance, ML algorithms have been successfully trained to distinguish MPs from organic debris in complex matrices using texture and spectral features.[61]

Another significant development is thermal extraction-desorption gas chromatography-mass spectrometry (TED-GC/MS), which allows for the analysis of larger environmental samples with minimal preparation. Unlike traditional Py-GC/MS, TED-GC/MS offers semi-quantitative analysis of polymer-specific signals without fully degrading the sample matrix.[62] Its high sensitivity and compatibility with multi-component detection make it ideal for low-concentration MPs in sediments and biota.

The emergence of bioresponsive sensors and aptamer-based platforms represents another frontier. These biosensors exploit molecular recognition properties to selectively bind polymer surfaces, allowing for rapid, label-free detection even in complex biological tissues.[63,64] SERS combined with nanostructured substrates is also being explored to detect nanoplastics down to ~100 nm, a scale beyond the resolution of conventional FTIR or Raman spectroscopy.[65]

Additionally, single-particle tracking techniques, such as nanoparticle tracking analysis (NTA) and flow cytometry adapted for MPs, are gaining attention.[66] These techniques enable real-time quantification and size distribution of small MPs and nanoplastics in aqueous suspensions, albeit with challenges in differentiating plastic from non-plastic particles without fluorescence tagging.

Despite these advances, several analytical gaps remain. First, no single method currently offers simultaneous high-throughput analysis, polymer identification, and nanoscale resolution. Second, matrix effects (*e.g.*, biogenic debris, organic matter) continue to hamper accuracy. Third, interlaboratory reproducibility is limited by the absence of validated reference materials and standardized protocols. Lastly, field-deployable, low-cost devices for real-time monitoring remain underdeveloped. Ongoing collaborative efforts—such as the European Commission's JRC harmonization initiatives and ISO standardization projects—are vital to address these gaps. The integration of complementary approaches and the adoption of novel tools from materials science, nanotechnology, and bioengineering will play a central role in advancing MPs analytics globally.

Case studies of MPs contamination in marine life

MPs pollution is a pervasive global issue that presents significant challenges to marine ecosystems and human health. Although MPs are ubiquitous in the world's oceans, their distribution, composition, and potential impacts vary widely among regions due to differences in plastic consumption patterns, waste management practices, and environmental policies. A series of recent case studies from diverse geographic locations illustrate these variances and shed light on the complexities of MPs contamination in marine life (Table 2). These studies not only document the presence of MPs in a range of marine organisms—from bivalve mollusks and fish to crustaceans and seaweed—but also provide insights into the sources, types, and potential risks associated with this pollution.

A number of investigations have focused on bivalve mollusks, which are especially vulnerable to MPs contamination due to their filter-feeding habits. In the Itapessoca estuary of Goiana, Pernambuco in Northeast Brazil, researchers examined the clam species *Anomalocardia flexuosa* during three different sampling periods (October 2019, December 2019, and February 2020). Their analysis revealed that 90% of the individuals examined were contaminated with MPs, averaging 5.15 ± 3.80 particles per individual and 3.66 ± 2.59 particles per gram of soft tissue. With fragments accounting for 54% of the detected particles and fibers 43%, the study not only marked the first record of MPs contamination in this clam species in South America but also demonstrated the suitability of *A. flexuosa* as a biomonitor for local MPs pollution.[29] The contamination in these clams is likely linked to local economic activities such as artisanal fishing, aquaculture, and tourism, which contribute various plastic wastes to the environment. Further studies on bivalves have been conducted in the coastal waters of Qingdao, China, where four species—scallops, mussels, oysters, and clams—were sampled across different seasons in 2018. The analysis employed both stereomicroscopy and ATR-μ-FTIR to identify and quantify the MPs. In this study, fibers were the most common type (45%), followed by films (28%), fragments (23%), and granules (4%). The particle size distribution indicated that over half of the MPs were smaller than 1000 μm, with contamination levels ranging between 0.5 and 3.3 items per individual. Notably, clams (*Ruditapes philippinarum*) exhibited significantly higher levels of contamination compared to other species, a result attributed to habitat differences that influence MPs ingestion. The study further suggested that local aquaculture equipment and urban wastewater discharges were likely sources of the plastics detected.[67] In the Mediterranean region, the Apulia region of Italy provided another compelling example of MPs contamination in bivalve mollusks. Sampling in fish markets around Bari and its neighboring towns between January and May 2022 focused on mussels (*Mytilus galloprovincialis*) and oysters (*Crassostrea gigas*). In mussels, fragments comprised approximately 60.96% of the particles while fibers accounted for 37.01%, with a predominance of polyamide (PA) and nylon 6/6 polymers. Oysters displayed a different profile, with a substantial presence of films (30.74%) alongside fragments and fibers. Although contamination varied by geographic origin, the overall size of MPs was most commonly between 5 and 500 μm, and the data suggested that factors related to the site of harvest were more influential than the species themselves. The diversity of potential sources in this region included aquaculture equipment and various land-based anthropogenic activities, underscoring the complex interplay between human activities and marine pollution.[68] Another extensive study from

Table 2 Summary of collected case studies on MPs contamination in marine life

Location	Sampling timeframe	Organisms studied	Sample size	Method of MPs analysis	Types of MPs	MPs size range	Level of contamination	Key findings	Potential sources	Ref.
Itapessoca estuary, Goiana, Pernambuco, Northeast Brazil	October 2019, December 2019, and February 2020	Bivalve mollusc: *Anomalocardia flexuosa* (clam species)	Total of 60 individuals	Stereomicroscope	Fragments: 54%, Fibers: 43%, Pellets: 3%	17 to 1057 µm	90% of individuals contained MPs; Average of 5.15 ± 3.80 MP particles per individual; 3.66 ± 2.59 particles per gram of soft tissue; Total of 309 MPs particles recovered from all samples; Range: 0–16 particles per individual	First record of MPs contamination in *A. flexuosa* in South America; Species proved suitable for monitoring MPs pollution levels; No correlation between number of MPs and clam biometry; Estimated consumer exposure: ~3660 MP particles per kg of clam meat	Local economic activities: artisanal fishing, aquaculture, and tourism; Anthropogenic influences from: Aquaculture operations, Real estate development, Tourism activities	29
Qingdao, China (specifically Huangdao (HD) and Shazikou (SZK) regions around Jiaozhou Bay)	November 2017 (autumn), February (winter), May (spring), and August (summer) 2018	Four bivalve species: Scallop (*Chlamys farreri*), Mussel (*Mytilus galloprovincialis*), Oyster (*Crassostrea gigas*), Clam (*Ruditapes philippinarum*)	290 bivalve samples (10 individuals per species)	Stereomicroscope + ATR-µ-FTIR	Fiber (45%), Film (28%), Fragment (23%), Granule (4%) PVC, Rayon, Cellophane, Polyester (PES), Chlorinated polyethylene, PET	>53% were smaller than 1000 µm; 27% were between 1000–2000 µm	Range: 0.5–3.3 items per individual; 0.3–20.1 items per g (w.w.), 80% detection rate (233 out of 290 samples)	Clams (*R. philippinarum*) contained significantly higher MPs levels than other species; Habitat type (sediment *vs.* water-dwelling) influenced MPs ingestion; No significant seasonal variation in abundance; Larger MPs found in summer and spring	Aquaculture industry (PVC buoys and tubes); Clothing and hygiene products (rayon); Agricultural plastic mulch film; Surface runoff during rainy seasons; Sewage discharge	67

Table 2 *(Contd.)*

Location	Sampling timeframe	Organisms studied	Sample size	Method of MPs analysis	Types of MPs	MPs size range	Level of contamination	Key findings	Potential sources	Ref.
Apulia region, Italy; Fish markets in Bari and neighboring towns	January to May 2022	Mussels (*Mytilus galloprovincialis*); Oysters (*Crassostrea gigas*)	Mussels: 180 individuals; Oysters: 18 individuals	Stereomicroscope + FTIR-ATR	Mussels: Fragments (60.96%), Fibers (37.01%), Films (0.38%), Spherical granules (2.3%); Oysters: Fragments (36.7%), Fibers (28.52%), Films (30.74%), Spherical granules (2.3%); Mussels: Primarily PA and nylon 6/6; Oysters: Primarily chlorinated PP	Overall range: 10 to 7350 μm; Most common size category: 5–500 μm (69.08% in mussels, 70.74% in oysters)	Mussels:0.6–1.9 items per g tissue; 2.5–6.6 items per individual; Total of 789 MPs detected; Oysters: 0.41–0.71 items per g tissue; 12–18.83 items per individual; Total of 270 MPs detected	Color distribution varied between species (mussels: predominantly blue; oysters: predominantly transparent); Statistical significance found between samples and procedural blanks; Site-related influence more important than species-related influence; Contamination levels varied by geographic origin	Aquaculture equipment (collectors, ropes, nets, tubes); Synthetic textile fibers from aquaculture operations; Marketing stage contamination; Environmental contamination from various anthropogenic activities; Land-based sources (plastic bags, packaging materials, industry waste)	68
Daya Bay, Southern China; Three inner bays studied: Dongshan (DS), Aotou (AT), and Fanhe (FH)	August (summer) and December (winter) 2019	*Chlamys nobilis* (scallop); *Perna viridis* (mussel); *Crassostrea hongkongensis* (oyster); *Paphia undulata* (clam); *Ruditapes philippinarum* (clam); *Meretrix meretrix* (clam)	Total of 540 individual bivalves analyzed; 15 individuals per species from each farming area	Stereomicroscope + μ-FTIR	Fibers (predominant – 86.5% in bivalves), Fragments, Films, Pellets; PET, PP, PE, PA, PS, PVC, PC, polyurethane, styrene-acrylonitrile, ethylene-vinyl acetate, polyoxymethylene, polyacrylonitrile, expanded polystyrene, polymethyl methacrylate	Majority <500 μm (>80% of total MPs in bivalves)	86.7% to 93.3% occurrence in different species; Abundance: 3.5 to 8.6 items per individual; Or 0.2 to 3.1 items per g tissues w.w.	Sediment-dwelling bivalves had higher MPs abundances than water-dwelling bivalves; No significant differences in MPs abundances across locations and seasons; Risk levels based on polymer hazard index (PHI) were II-III for most species; MPs features in bivalves were similar to those in their ambient environment	Municipal waste; Sewage discharge; Industrial effluents; Textile industry (especially for fibers); Aquaculture activities; Shipping; Tourism activities	69

Table 2 *(Contd.)*

Location	Sampling timeframe	Organisms studied	Sample size	Method of MPs analysis	Types of MPs	MPs size range	Level of contamination	Key findings	Potential sources	Ref.
Coastal areas of Thailand along the Gulf of Thailand: Chonburi, Samut Sakhon, Samut Songkhram, Phetchaburi, Chumphon, and Surat Thani	August 2020 to March 2021	Green mussels (*Perna viridis*); Oysters (*Saccostrea cucullata*); Cockles (*Tegillarca granosa*); Clams (*Meretrix meretrix*)	30 individuals of each species at each location; total of 562 potential MPs items analyzed	Stereomicroscope + FTIR-ATR	Fibers (95.72%), Fragments (2.50%), Granules (1.78%); Cotton (46.76%), PET (35.54%), Rayon (8.33%), PP (4.75%), PE (1.68%), Acrylic (1.59%), PA (0.68%), PS (0.67%)	≤100 µm (15.15%); >100–500 µm (62.66%); >500–1000 µm (16.31%); >1000–5000 µm (5.88%)	Detection rate: 69% to 93% (average 74.83%); Average abundance: 1.87 ± 0.86 items per individual or 0.46 ± 0.43 items per g w.w.; Range: 0 to 16 items per individual	Benthic bivalves (cockles and clams) contained more MPs than pelagic species (mussels and oysters); Small blue microfibers (<500 µm) were most abundant; Natural-based polymers dominated (cotton and rayon); Daily MPs exposure for consumers: 0.52 items per person; Low to moderate health risk identified for Thai bivalve consumers	Textile industry waste; Laundry water discharges; Wastewater treatment plants; Aquaculture equipment (blue plastic mesh bags, ropes, floating barrels); River inputs and municipal drainage systems; Land-based sources (urbanization, industrial development, tourism);	70
Kuakata, Bangladesh, Bay of Bengal (North Bay of Bengal)	September 2019	10 commercial marine fish species: Demersal species: *Priacanthus hamrur, Sciades sona, Carangoides chrysophrys*; Benthopelagic species: *Harpadon nehereus, Otolithoides pama*; Pelagic-neritic species: *Setipinna tenuifilis, Coilia neglecta, Anodontostoma chacunda, Sardinella brachysoma*; Pelagic species: *Megalaspis cordyla*	Total 100 individual fish (10 individuals per species)	Stereomicroscope + SEM + FTIR-ATR	Fibers (53.4%), Films (40%), Fragments (3.3%), Foams (1.9%), Granules (1.4%); PE (55%) PP (33%), PES (6%), polyurethane (PU) (2%), EPDM (2%)	Three size classes: <500 µm (85% of particles), >500 to <1000 µm, >1000 to <5000 µm	Total MPs found: 215 particles; Average: 2.2 ± 0.89 items per individual; Range: 1.0–3.8 items per species; Concentration: 0.02–0.15 MPs per gBW and 0.45–6.45 MPs per gGIT	All studied species contained MPs; Higher abundance in demersal fish compared to pelagic species; Green colored MPs were most prevalent (39%); Smaller MPs (<500 µm) dominated; Surface morphology showed signs of weathering	Domestic laundry discharge through wastewater treatment plants; Fragmentation of fishing gear (ropes and nets); Single-use plastics; Improper waste management along the Bay of Bengal basin; Aquaculture industry	71

Table 2 *(Contd.)*

Location	Sampling timeframe	Organisms studied	Sample size	Method of MPs analysis	Types of MPs	MPs size range	Level of contamination	Key findings	Potential sources	Ref.
Southern New Zealand (within 12 nautical miles of the coast from Oamaru to Te Waewae Bay)	2019–2020 (samples collected over multiple months throughout all seasons)	10 commercial fish species including: Red Cod (*Pseudophycis bachus*), Tarakihi (*Nemadactylus macropterus*), Red Gurnard (*Chelidonichthys kumu*), Butterfly Perch (*Caesioperca lepidoptera*), Sea Perch (*Helicolenus barathri*), Blue Moki (*Latridopsis ciliaris*), Spiny Dogfish (*Squalus acanthias*), Barracouta (*Thyrsites atun*), Jack Mackerel (*Trachurus declivis*), Blue Warehou (*Seriolella brama*)	155 fish	Stereomicroscope + Raman	Fibers (most dominant: 86.7% in pelagic, 82.3% in benthic), Fragments, Films, Beads, Bundles; PE (68%), Viscose (14%), PE + plumbophyllite (12%), PP + phthalocyanine (2%), PP + TiO_2 (2%), PE + hydroxy-apatite (2%)	99.68% <5 mm; 98.05% <3 mm	75% of fish contained MPs; Average: 2.5 particles per fish; Total: 391 MPs pieces found	High contamination rate compared to similar Australian studies; No significant difference between benthic and pelagic fish ingestion; Blue, black, and red were most common colors; Most particles were smaller than 3 mm	Sewage outflow (particularly for fibers); Wastewater treatment plants; Fishing gear (especially blue PE rope); Textile washing; Industrial discharge	35
Malaysia: Pantai Remis, Selangor, Mersing, Johor	Not explicitly stated	16 species including: Wolf herring, Band sickelfish, Spotted batfish, Jones' pony fish, Slimy pony fish, Deep body mojarra, Sardine, Silver tripod fish, Longtail tripod fish, Bumpnosed trevally, Yellowtail scad, Jack mackeral, Hammer croaker, Panna croaker, Indian threadfish, Hardtail scad	158 fishes	Stereomicroscope + FTIR-ATR + Raman + SEM-EDX	Fibers (80.2%), Fragments (17.7%), Filaments (3.1%); PA, PET, PP, PE, PS, ABS	0.063–0.1 mm (4%), 0.1–0.5 mm (36.3%), 0.5–1.0 mm (31.9%), 1.0–5.0 mm (27.7%)	86% of fish contained MPs in GIT; 92% contained MPs in gills; Average per individual: Pantai Remis: 9.88 items per individual; Mersing: 5.17 items per individual	Higher MPs occurrence in fish from urban areas; Larger MPs concentrated in gills; Smaller MPs dominated in GIT; Heavy metals (Cr and Fe) detected on colored MPs; Widespread distribution in commercial marine fishes; Potential human exposure through fish consumption	Urban waste and effluents; Tourism and recreational activities; Fishing activities and lost fishing gear; Landfill leachate through river systems; Textile fibers from domestic discharge; Industrial activities	34

Table 2 *(Contd.)*

Location	Sampling timeframe	Organisms studied	Sample size	Method of MPs analysis	Types of MPs	MPs size range	Level of contamination	Key findings	Potential sources	Ref.
Haizhou Bay, China	October 2020	Three types of fish: Pelagic fish (*e.g.*, *Konosirus punctatus, Ilisha elongata*); Demersal fish (*e.g.*, *Pampus argenteus, Collichthys lucidus*); Benthic fish (*e.g.*, *Cynoglossus joyneri, Tridentiger barbatus*)	146 fish	Stereomicroscope + FTIR	Fibers (dominant in all media), Fragments, Granules, Foam, PET, PE, PVC	Majority were <1 mm; Some particles >5 mm (1.1% in fish)	Pelagic fish: 2.15 ± 1.55 items per fish; Demersal fish: 2.88 ± 3.12 items per fish; Benthic fish: 2.82 ± 2.60 items per fish	MPs more abundant in bottom/middle water than surface water; Demersal and benthic fish contained more MPs than pelagic fish; Evidence of trophic transfer (TMF = 1.46); Body length and weight significantly affected MP ingestion; Strong correlation between MPs in pelagic fish and surface water	Domestic wastewater discharge; Fishing operations (nets, lines, gear); Textile industry; Packaging materials; Personal care products; River inputs (80% of MP pollutants); Marine activities (20% of MPs)	72
Giresun province, Southern Black Sea coast, Türkiye	September 20, 2022	Four fish species: Pelagic species: European anchovy (*Engraulis encrasicolus*), Horse mackerel (*Trachurus trachurus*); Benthic species: Red mullet (*Mullus barbatus*), Whiting (*Merlangius merlangus*)	40 fish	Stereomicroscope + FTIR-ATR	Fibers (dominant type: 47–82%), Fragments, Pellets; PP (56%), PA/nylon (25%), PES (19%)	0–0.5 mm: 16%; 0.5–1 mm: 26%; 1–2 mm: 28%; 2–5 mm: 29%	Overall mean: 1.7 ± 0.18 items per fish; Engraulis encrasicolus: 1.8 items per fish; Trachurus trachurus: 1.6 items per fish; Mullus barbatus: 1.9 items per fish; Merlangius merlangus: 1.8 items per fish	MPs present in all examined species; Color distribution: Black (41%), Blue (24%), Red (24%), Green (11%); No significant difference in MP ingestion between pelagic and demersal species; Fiber was the most common type, highest in anchovy (82%); Potential threat to human food safety even if GIT is removed before cooking	Clothing and fishing nets (fiber-type MPs); River inputs (Aksu and Pazarsuyu Rivers); Tourism activities; International port activities; Small-scale industries; Busy Black Sea international highway; Plastic bags, bottles, and packaging materials	73

Table 2 *(Contd.)*

Location	Sampling timeframe	Organisms studied	Sample size	Method of MPs analysis	Types of MPs	MPs size range	Level of contamination	Key findings	Potential sources	Ref.
Five Australian states/territories: Western Australia (WA), South Australia (SA), New South Wales (NSW), Queensland (QLD), Northern Territory (NT)	June 2019 to May 2020	Five species of decapod crustaceans: Three prawn species: king prawns (*Melicertus* spp.), tiger prawns (*Penaeus esculentus*), banana prawns (*Penaeus merguiensis*); Two crab species: blue-swimmer crab (*Portunus armatus*), mud crab (*Scylla serrata*)	510 specimens	Stereomicroscope + μ-FTIR	99% fibers, 1.4% fragments; Polyolefins (28%), PES(21%), PE(15%)	78.5% between 38 μm and 1 mm; 21.5% greater than 1 mm	Overall frequency: 48% of all crustaceans contained MPs; Mean abundance: 0.98 ± 0.06 items per individual; Species-specific: Prawns: 0.8 ± 0.1 items per individual, Crabs: 1.6 ± 0.1 items per individual, Highest in mud crabs (80% occurrence)	Contamination levels lower than global average but comparable to similar methodological studies; Regional variations in contamination levels; Larger species (mud crabs) showed higher contamination rates; NT showed highest contamination levels for several species	Land-based sources (*e.g.*, synthetic clothing fibers *via* washing machines); Fisheries equipment degradation; Ocean currents transporting plastic waste; Wastewater discharges	74
Five commercial prawn grounds around west and northeast coast of Ireland: Aran Prawn Ground (APG), Magharees Union (MU), Kenmare Bay (KB), Bantry Bay (BB), North Irish Sea (NIS)	November-December 2016	Nephrops norvegicus (Dublin Bay Prawn/Norway Lobster)	186 individuals	FTIR-ATR	Fibers (98.1%), Fragments (1.5%), Films (0.4%); PS, PP, PES, PE, PVC, PA	143.20 μm to 16.976 mm; Most common size class: 1–2 mm (32.0%); 86.2% within MPs definition (<5 mm)	Average: 1.75 ± 2.01 items per individual; 69% of samples (103 out of 150) contained at least 1 MPs particle; Range: 1–10 items per individual	Highest contamination in Kenmare Bay (2.30 ± 2.47 items per individual); Lowest in Aran Prawn Grounds (0.90 ± 1.03 items per individual); Positive correlation between MPs ingestion and carapace length; Estimated human exposure: 15–4471 particles per year through prawn consumption	Local fishing gear (nets and floats); Polymer types match commonly used fishing equipment; Environmental degradation of larger plastic items	75

Table 2 *(Contd.)*

Location	Sampling timeframe	Organisms studied	Sample size	Method of MPs analysis	Types of MPs	MPs size range	Level of contamination	Key findings	Potential sources	Ref.
Southern coast of the Caspian Sea, Iran	June to August 2023	Gammarus (*Pontogammarus maeoticus*), Shrimp (*Palaemon elegans*)	30 specimens of *P. maeoticus* per station; 15 specimens of *P. elegans* per station	Stereomicroscope + Raman	Fibers (85.85%), Fragments (10.61%), Films (3.54%); PA (61.54%), PC (23.07%), PU (11.54%), PVDF (3.85%)	30–5000 µm overall; Most common: 300–1000 µm (32.78%); *P. elegans*: 596.62 µm (65–2057 µm); *P. maeoticus*: 293.82 µm (30–535 µm)	*P. elegans*: 0.38 ± 0.21 items per individual or 0.58 ± 0.34 items per g w.w.; *P. maeoticus*: 0.26 ± 0.15 items per individual or 8.69 ± 7.88 items per g w.w.	Both studied organisms showed higher bioconcentration from water than sediment; No correlation between MPs abundance in organisms and their wet weight; *P. maeoticus* and *P. elegans* were found unsuitable as biomonitors for MPs pollution	Fishing gear (nets, lines, ropes); Domestic sewage (especially laundry washing); Urban surface runoff; Tourism activities; Transportation; Improper disposal of household and industrial waste	76
East Asia; China (Changdao, Rongcheng, Zhoushan, Fangchenggang, Quanzhou, Lianyungang); South Korea (Cheongju); Japan (Saga); North Korea (northern coast)	2019	Brown seaweed (*Saccharina japonica*); Red seaweed (*Pyropia yezoensis*)	11 commercially available seaweed samples: 7 varieties of *Saccharina japonica*; 4 varieties of *Pyropia yezoensis*	Stereomicroscope + µ-FTIR	Fibers (90–91% predominant), Fragments (9%), Films (1%); Cellophane (67%), PET (14%), PE (12%), PA (7%)	Most common size ranges: 100–500 µm (48.3%), 500–2000 µm (44.2%); *S. japonica*: 436 µm, *P. yezoensis*: 856 µm	*S. japonica*: 2.3–12.7 items per g d.w; *P. yezoensis*: 2.9–5.0 items per g d.w.	Seaweeds contribute up to 45.5% of dietary MPs intake in East Asia; Chinese individuals consume estimated 17 034 MPs per person per year through seaweed; Conventional washing methods remove <38% of MPs; Higher contamination in Chinese samples compared to Japan, Korea, and North Korea; Women face higher risk of MPs ingestion than men	Marine environment contamination; Processing and packaging materials; Aquaculture activities; Fishing gear degradation; Atmospheric deposition	77

Table 2 *(Contd.)*

Location	Sampling timeframe	Organisms studied	Sample size	Method of MPs analysis	Types of MPs	MPs size range	Level of contamination	Key findings	Potential sources	Ref.
Mar Piccolo of Taranto (Ionian Sea, Mediterranean Sea, Southern Italy); IMTA system located in Mar Grande, about 600 m from coast	Samples collected during period of maximum growth (July); Seaweed cultivated for six months in the IMTA system	Green seaweed Chaetomorpha linum (O.F. Müller) Kützing (Chlorophyta, Cladophorales)	Three replicates of about 500 g of *C. linum*	FTIR-ATR	Fibers (237 pieces, 97.3%); Fragments (4 pieces); Films (4 pieces)	0.025 mm to 1.99 mm; Most prevalent size class: 0.80–1.00 mm	245 MPs retrieved from the samples	*C. linum* effectively traps MPs from marine environment; MPs can be successfully removed from harvested algae using NaCl solution washing; The seaweed can serve dual purpose: as MP bioremediator and source of bioactive compounds; Estimated production of 0.84t algal biomass per annual cycle in the IMTA system	Aquaculture equipment (boats, fish cages, buoys, nets, ropes); Textile industry (synthetic fibers from washing machine effluent); Plastic bags and packaging; Maritime activities (military and commercial vessels); Industrial activities in Taranto (steel factories, oil refineries, chemical plants)	78

Daya Bay in Southern China evaluated several bivalve species, including scallops, mussels, oysters, and clams, across two seasons (summer and winter 2019). This investigation revealed that fibers were predominant (86.5% in bivalves) and that more than 80% of the MPs were smaller than 500 μm. The overall occurrence of MPs ranged from 86.7% to 93.3% among different species, with an abundance between 3.5 and 8.6 items per individual. Notably, sediment-dwelling bivalves exhibited higher levels of MPs contamination than their water-dwelling counterparts. The study also found that the polymer hazard index (PHI) for most species fell within moderate risk levels (II–III), suggesting that the MPs features in these organisms closely mirrored those in their surrounding environment. The authors attributed the contamination primarily to municipal waste, sewage discharge, industrial effluents, and local aquaculture practices.[69] Similarly, research conducted in coastal areas of Thailand along the Gulf of Thailand from August 2020 to March 2021 examined green mussels, oysters, cockles, and clams. This study reported an average detection rate of approximately 74.83% for MPs, with fibers overwhelmingly dominant at 95.72% of the total count. The majority of particles were within the 100–500 μm range, and benthic species such as cockles and clams were found to have higher MPs levels compared to pelagic species. Textile industries, wastewater discharges, and aquaculture gear contribute to MPs. These raise concerns about food safety even at moderate exposure levels.[70]

Fish, as an integral part of the marine food web and an important human dietary resource, have also been extensively studied for MPs contamination. In the Bay of Bengal near Kuakata, Bangladesh, a study encompassing 10 commercial marine fish species—including demersal, benthopelagic, and pelagic species—demonstrated that all species contained MPs, with an overall average of 2.2 ± 0.89 items per individual. The study highlighted that demersal fish exhibited higher MPs abundance compared to their pelagic counterparts. Fibers, films, and fragments were detected in various proportions, with smaller particles (<500 μm) dominating the samples. These findings pointed to domestic laundry discharges, fishing gear fragmentation, and improper waste management as likely sources of the MPs observed.[71] In Southern New Zealand, a comprehensive study evaluated 10 commercial fish species collected over multiple seasons from coastal waters. Using a combination of stereomicroscopy and Raman spectroscopy, the study found that 75% of the sampled fish contained MPs, with an average of 2.5 particles per individual and a total of 391 MPs pieces recovered. The particles were predominantly fibers, with most being smaller than 3 mm. While no significant differences in MPs ingestion were observed between benthic and pelagic fish, the high overall contamination rate suggested that local sources, such as sewage outflow and fishing gear degradation, were significant contributors to the MPs burden in these waters.[35] The situation in Malaysia further underscores the potential human exposure to MPs through fish consumption. In a study conducted along several coastal locations, 158 fish representing 16 species were analyzed using an array of advanced spectroscopic techniques. The results revealed that 86% of the fish contained MPs in their gastrointestinal tracts and 92% in their gills. The average number of MPs items per individual varied among sampling sites, with urban areas showing higher levels of contamination. The detected MPs, which ranged from 0.063 to 5.0 mm, were predominantly fibers and were frequently associated with heavy metals such as chromium and iron. This finding highlighted the potential for chemical co-contaminants to interact with MPs, thus exacerbating the risks posed to human health through seafood consumption.[34] A further study from Haizhou Bay, China, investigated three categories of fish—pelagic, demersal, and benthic—by examining 146 individuals using stereomicroscopy coupled with FTIR analysis. The study observed that while fibers were dominant in all fish types, demersal and benthic species contained slightly higher levels of MPs compared to pelagic species. With a reported trophic transfer factor (TMF) of 1.46, the study underscored that MPs transfer through the food web is a real and measurable phenomenon. Moreover, body length and weight were found to have significant correlations with MPs ingestion, and a strong association was noted between MPs in pelagic fish and those present in surface water, further linking marine contamination to local anthropogenic inputs such as domestic wastewater discharge and packaging materials.[72] In the Black Sea region, a study conducted along the Southern coast of the Giresun province in Türkiye evaluated four fish species, including both pelagic and benthic types. The findings indicated that all examined species contained MPs, with fibers being the most prevalent. Although the average MPs load per fish was relatively low (approximately 1.7 items per individual), the study raised concerns about the potential for human exposure, particularly since many of the particles were found to be of sizes that might not be completely removed by typical cleaning practices.[73]

Crustaceans have not been overlooked in recent research. In a broad survey covering five Australian states and territories, 510 specimens of decapod crustaceans—including three prawn species and two crab species—were analyzed for MPs contamination. The study revealed that nearly 99% of the MPs found were fibers, with contamination frequencies varying by species; for instance, mud crabs exhibited the highest occurrence rate of MPs ingestion. The research attributed the plastic contamination largely to land-based sources, such as synthetic clothing fibers from domestic washing, as well as the degradation of fishing gear and the influence of ocean currents in transporting plastic waste.[74] A complementary study from Ireland focused on the commercially valuable Nephrops norvegicus (Dublin Bay prawn or Norway lobster) across five prawn grounds. With an average of 1.75 items per individual and a positive correlation between MPs ingestion and carapace length, the study underscored that even after standard cleaning procedures such as deveining, consumers could be exposed to MPs originating from both local fishing gear and broader environmental sources.[75] In the southern coast of the Caspian Sea in Iran, a more recent investigation examined both gammarids and shrimp. Although these organisms exhibited relatively low levels of contamination, the study noted that the MPs characteristics—predominantly fibers with a range of sizes—were reflective of inputs from fishing gear and domestic sewage. Interestingly, the researchers concluded that these species might not serve well as biomonitors for MPs pollution due to the lack of a consistent correlation between organism wet weight and MPs abundance.[76]

Beyond animal species, studies have also focused on seaweeds, which play a dual role as both food and potential bioindicators of marine pollution. Research across East Asia, encompassing China, South Korea, Japan, and North Korea, analyzed brown seaweed (*Saccharina japonica*) and red seaweed (*Pyropia yezoensis*) for MPs contamination. The results showed an overwhelming predominance of fibers, with cellophane, polyethylene terephthalate (PET), and PE being the most common polymers detected. The study estimated that seaweed contributed up to 45.5% of the dietary MPs intake in the region, with some estimates suggesting that Chinese individuals might ingest as many as 17 034 MPs particles per year through seaweed consumption. These findings have serious implications for food safety, as conventional washing methods appear to remove less than 38% of the MPs present on the algae.[77] In another innovative study from the Mar Piccolo of Taranto in Southern Italy, researchers examined the cultivation of the green seaweed *Chaetomorpha linum* within an integrated multi-trophic aquaculture (IMTA) system. The study found that the seaweed not only accumulated significant quantities of MPs—primarily fibers—but also demonstrated the potential to act as a bioremediator, with the possibility of removing trapped MPs through washing with a sodium chloride solution. This dual role underscores the potential for certain seaweeds to contribute to both food production and environmental cleanup efforts, although the presence of MPs in the harvested biomass still poses concerns for human exposure and product safety.[78]

Collectively, these case studies from disparate geographic regions offer a comprehensive view of the current status of MPs contamination in marine life. The studies highlight how variations in local industrial practices, urbanization, and even seasonal factors can influence the degree and types of MPs present in marine organisms. In bivalve mollusks, the contamination levels appear to be influenced largely by local environmental conditions and aquaculture practices, whereas in fish and crustaceans, feeding behavior, habitat, and trophic dynamics play crucial roles in determining MPs ingestion rates. Moreover, the detection of MPs in seaweeds extends the concern to primary producers and dietary staples in certain regions, further complicating the risk assessment for human consumers. The cumulative evidence underscores the need for enhanced monitoring and improved waste management strategies that address the diverse and complex sources of MPs. As research continues to evolve, it remains imperative that future studies focus on the long-term ecological and human health implications of chronic MPs exposure, while also exploring innovative remediation strategies to mitigate this pervasive pollutant.

Conclusion and perspectives

MPs pollution has firmly been established as a contaminant of concern in marine ecosystems worldwide. Our review highlights that MPs are present in all marine habitats and have been found in a vast array of organisms, from plankton and corals to fish and marine mammals. There is clear evidence that MPs negatively affect marine life – over 90% of studied species show some form of impact ranging from mild physiological stress to severe injury or death under high exposure scenarios. MPs also permeate the seafood we consume, introducing a direct pathway for human exposure. We have reviewed how different detection methods reveal complementary aspects of the problem: visual and spectroscopic techniques show the number and types of MPs organisms are ingesting, while bulk chemical analyses confirm that a broad spectrum of polymers have entered marine food webs.

Despite the advances in understanding, important gaps remain. One is the human health impact. While it is intuitive that consuming plastics is undesirable, we lack conclusive data on how chronic dietary MPs exposure affects human health. Toxicological studies are needed to determine if MPs could cause subtle long-term effects such as gut microbiome changes, inflammation, or uptake of harmful substances in people. Epidemiological research could also investigate correlations between high seafood consumption and any health markers related to MPs. Another gap is in the very small particle range: nanoplastics. Current analytical methods struggle to detect nanoparticles, yet these could be even more bioavailable and toxic at the cellular level than larger MPs. Developing reliable methods to measure nanoplastics in environmental and biological samples is a frontier challenge.

As MPs become increasingly prevalent in marine organisms and seafood, concerns about their implications for human health have intensified. Humans are exposed to MPs through multiple pathways, most notably by consuming contaminated seafood, seaweed, and table salt, and through the ingestion of particles present in drinking water. Inhalation of airborne MPs and, to a lesser extent, dermal contact through personal care products or marine recreation may also contribute to exposure. While direct causal links between MPs ingestion and specific human health outcomes remain unconfirmed, recent studies suggest that MPs can cross biological barriers, accumulate in tissues, and potentially induce oxidative stress, inflammation, and disruption of the gut microbiome. The discovery of plastic particles in human blood and lung tissues underscores the urgency of further toxicological and epidemiological investigations into their health effects. Despite growing evidence of widespread human exposure, regulatory frameworks remain limited and fragmented. The European Food Safety Authority (EFSA) has acknowledged the presence of MPs in food but concluded that existing data are insufficient to establish a health-based guidance value. Similarly, the World Health Organization (WHO) has urged further research before setting regulatory thresholds, emphasizing that current knowledge does not allow a definitive risk assessment. At present, no binding legal limits for MPs in food, beverages, or environmental media exist under national or international law, which hampers coordinated risk management efforts.

To bridge this gap between emerging science and regulatory action, we propose a conceptual framework to guide the development of standardized protocols and threshold-based risk assessments. The framework begins with the harmonization of sampling, isolation, and analysis techniques to ensure data comparability across studies and regions. It recommends the adoption of unified reporting metrics, such as particle concentration per gram or milliliter and polymer-specific mass concentrations, to improve exposure characterization. It calls for long-term toxicological studies to establish realistic dose–response relationships under environmentally relevant conditions. These data can, in turn, inform the development of provisional tolerable daily intake (PTDI) levels for key polymer types. Finally, this evidence base should be integrated into policy instruments through collaborative international efforts, including standard-setting bodies like the ISO, regional marine conventions, and emerging agreements such as the United Nations global treaty on plastic pollution. Advancing along this trajectory will ensure that MPs research is aligned with regulatory objectives, thereby enabling effective protection of public health and the marine environment.

There is also a need for standardization in MPs research methods. Different studies use different sampling nets, digestion procedures, and identification criteria, which can lead to highly variable results. Efforts are underway to create standard protocols (*e.g.*, through organizations like ISO and regional sea conventions) so that data from different regions are more comparable. Standard reference materials (like synthetic sediment or biota spiked with known MPs) could help laboratories validate their methods. Moreover, long-term monitoring programs are still in their infancy for MPs. For MPs already in the ocean, mitigation is tricky, but some local efforts are possible. Cleaning beaches and coastal areas can at least remove plastic before it breaks down into MPs. There are also experimental efforts to collect MPs from surface waters using special skimmers or filtration boats, though scaling that up is daunting given the vast ocean area.

To effectively control future inputs of MPs into marine ecosystems, a multipronged strategy is essential. First, source reduction through bans or restrictions on single-use plastics, especially those most commonly found in marine environments (*e.g.*, polyethylene bags, polystyrene foam containers), should be prioritized. Second, improved waste management infrastructure, including recycling programs and advanced filtration in wastewater treatment plants (*e.g.*, membrane bioreactors), can significantly reduce plastic leakage into aquatic systems. Third, industrial regulations must be enforced to limit the release of microbeads and microfibers from manufacturing and textile sectors, potentially through technologies such as microfiber filters in washing machines or the adoption of biodegradable materials. Fourth, public education and behavioral change campaigns are necessary to raise awareness about plastic pollution and promote sustainable consumption practices. Finally, international cooperation and policy harmonization are vital, as marine plastic pollution is a transboundary issue. Global agreements, such as the UN Plastics Treaty currently under negotiation, could play a crucial role in setting binding targets and standardizing reporting and monitoring protocols. Collectively, these interventions can help reduce the influx of MPs into marine systems and mitigate their long-term ecological and human health impacts.

Author contributions

Hassan Karimi-Maleh: conceptualization, supervision, methodology, funding acquisition, project administration. Li Fu: conceptualization, supervision, methodology, writing – review & editing. Qingwei Zhou: investigation, formal analysis, data curation, visualization. Meiqing Jin: investigation, formal analysis, data curation. Weihong Wu: investigation, data curation. Cheng-Te Lin: supervision, writing – review & editing.

Conflicts of interest

There are no conflicts to declare.

Data availability

No primary research results, software or code have been included and no new data were generated or analysed as part of this review.

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References

1 M. Cole, P. Lindeque, C. Halsband and T. S. Galloway, *Mar. Pollut. Bull.*, 2011, **62**, 2588–2597.
2 R. C. Thompson, Y. Olsen, R. P. Mitchell, A. Davis, S. J. Rowland, A. W. G. John, D. McGonigle and A. E. Russell, *Science*, 2004, **304**, 838–838.
3 J. R. Jambeck, R. Geyer, C. Wilcox, T. R. Siegler, M. Perryman, A. Andrady, R. Narayan and K. L. Law, *Science*, 2015, **347**, 768–771.
4 A. L. Andrady, *Mar. Pollut. Bull.*, 2011, **62**, 1596–1605.
5 P. J. Kershaw and C. M. Rochman, Reports and studies-IMO/FAO/Unesco-IOC/WMO/IAEA/UN/UNEP joint group of experts on the scientific aspects of marine environmental protection (GESAMP) Eng No. 93.
6 K. Mattsson, S. Jocic, I. Doverbratt and L.-A. Hansson, in *Microplastic Contamination in Aquatic Environments*, ed. E. Y. Zeng, Elsevier, 2018, pp. 379–399.
7 A. L. Lusher, V. Tirelli, I. O'Connor and R. Officer, *Sci. Rep.*, 2015, **5**, 14947.
8 M. A. Browne, A. Dissanayake, T. S. Galloway, D. M. Lowe and R. C. Thompson, *Environ. Sci. Technol.*, 2008, **42**, 5026–5031.
9 L. C. Woodall, A. Sanchez-Vidal, M. Canals, G. L. J. Paterson, R. Coppock, V. Sleight, A. Calafat, A. D. Rogers, B. E. Narayanaswamy and R. C. Thompson, *R. Soc. Open Sci.*, 2014, **1**, 140317.
10 L. M. Rios, C. Moore and P. R. Jones, *Mar. Pollut. Bull.*, 2007, **54**, 1230–1237.
11 J. R. Jambeck, R. Geyer, C. Wilcox, T. R. Siegler, M. Perryman, A. Andrady, R. Narayan and K. L. Law, *science*, 2015, **347**, 768–771.
12 M. Eriksen, L. C. M. Lebreton, H. S. Carson, M. Thiel, C. J. Moore, J. C. Borerro, F. Galgani, P. G. Ryan and J. Reisser, *PLoS One*, 2014, **9**, e111913.
13 A. L. Lusher, M. McHugh and R. C. Thompson, *Mar. Pollut. Bull.*, 2013, **67**, 94–99.
14 S. L. Wright and F. J. Kelly, *Environ. Sci. Technol.*, 2017, **51**, 6634–6647.
15 M. Smith, D. C. Love, C. M. Rochman and R. A. Neff, *Curr. Environ. Health Rep.*, 2018, **5**, 375–386.
16 L. Van Cauwenberghe and C. R. Janssen, *Environ. Pollut.*, 2014, **193**, 65–70.
17 T. S. Galloway, M. Cole and C. Lewis, *Nat. Ecol. Evol.*, 2017, **1**, 1–8.
18 S. Rist, B. Carney Almroth, N. B. Hartmann and T. M. Karlsson, *Sci. Total Environ.*, 2018, **626**, 720–726.
19 R. Akhbarizadeh, F. Moore and B. Keshavarzi, *Food Addit. Contam.: Part A*, 2019, **36**, 1696–1708.
20 A. C. Vivekanand, S. Mohapatra and V. K. Tyagi, *Chemosphere*, 2021, **282**, 131151.
21 V. Hidalgo-Ruz, L. Gutow, R. C. Thompson and M. Thiel, *Environ. Sci. Technol.*, 2012, **46**, 3060–3075.
22 L. G. A. Barboza and B. C. G. Gimenez, *Mar. Pollut. Bull.*, 2015, **97**, 5–12.
23 Z. Huang, B. Hu and H. Wang, *Environ. Chem. Lett.*, 2023, **21**, 383–401.
24 F. Pfeiffer and E. K. Fischer, *Front. Environ. Sci.*, 2020, **8**, 572424, DOI: **10.3389/fenvs.2020.572424**.
25 EFSA Panel on Contaminants in the Food Chain (CONTAM), *EFSA J.*, 2016, **14**, e04501.
26 A. Dehaut, A.-L. Cassone, L. Frère, L. Hermabessiere, C. Himber, E. Rinnert, G. Rivière, C. Lambert, P. Soudant, A. Huvet, G. Duflos and I. Paul-Pont, *Environ. Pollut.*, 2016, **215**, 223–233.
27 X. Li, L. Fu, F. Chen, Y. Lv, R. Zhang, S. Zhao and H. Karimi-Maleh, *Anal. Methods*, 2025, **17**, 4300–4320, DOI: **10.1039/D5AY00612K**.
28 A. Dehaut, L. Hermabessiere and G. Duflos, *TrAC, Trends Anal. Chem.*, 2019, **116**, 346–359.
29 D. N. A. Bruzaca, A. K. S. Justino, G. C. P. Mota, G. A. Costa, F. Lucena-Frédou and A. O. Gálvez, *Mar. Pollut. Bull.*, 2022, **179**, 113659.
30 H. Woo, K. Seo, Y. Choi, J. Kim, M. Tanaka, K. Lee and J. Choi, *Appl. Sci.*, 2021, **11**, 10640.
31 P. Zhou, K. Zhang, T. Zhang, C. Cen, Y. Zheng and Y. Shuai, *Water*, 2024, **16**, 1898.

32 M. Khoshmanesh, A. M. Sanati, S. Shahcheragh, S. Farjadfard, Z. Bonyadi and B. Ramavandi, *Arabian J. Chem.*, 2024, **17**, 105737.

33 N. Saha and A. Shourie, *Environ. Qual. Manage.*, 2024, **34**, e22267.

34 N. Jaafar, A. Azfaralariff, S. M. Musa, M. Mohamed, A. H. Yusoff and A. M. Lazim, *Sci. Total Environ.*, 2021, **799**, 149457.

35 I. K. Clere, F. Ahmmed, P. I. J. G. Remoto, S. J. Fraser-Miller, K. C. Gordon, V. Komyakova and B. J. M. Allan, *Mar. Pollut. Bull.*, 2022, **184**, 114121.

36 W. Dellisanti, M. M.-L. Leung, K. W.-K. Lam, Y. Wang, M. Hu, H. S. Lo and J. K. H. Fang, *Mar. Pollut. Bull.*, 2023, **186**, 114221.

37 J. C. Prata, J. P. Da Costa, A. C. Duarte and T. Rocha-Santos, *TrAC, Trends Anal. Chem.*, 2019, **110**, 150–159.

38 B. N. Vinay Kumar, L. A. Löschel, H. K. Imhof, M. G. J. Löder and C. Laforsch, *Environ. Pollut.*, 2021, **269**, 116147.

39 J. Wagner, Z.-M. Wang, S. Ghosal, C. Rochman, M. Gassel and S. Wall, *Anal. Methods*, 2017, **9**, 1479–1490.

40 G. R. Bitencourt, P. A. Mello, E. M. M. Flores, C. Pirola, D. Carnaroglio and C. A. Bizzi, *Sci. Total Environ.*, 2020, **749**, 142301.

41 M. M.-L. Leung, Y.-W. Ho, C.-H. Lee, Y. Wang, M. Hu, K. W. H. Kwok, S.-L. Chua and J. K.-H. Fang, *Environ. Pollut.*, 2021, **289**, 117648.

42 M. M.-L. Leung, Y.-W. Ho, E. A. Maboloc, C.-H. Lee, Y. Wang, M. Hu, S.-G. Cheung and J. K.-H. Fang, *J. Hazard. Mater.*, 2021, **420**, 126541.

43 I. A. Guschina, A. J. Hayes and S. J. Ormerod, *Environ. Pollut.*, 2020, **263**, 114425.

44 M. Majewsky, H. Bitter, E. Eiche and H. Horn, *Sci. Total Environ.*, 2016, **568**, 507–511.

45 M. Shunmughanathan and K. Pitchumani, in *Polymers and Multicomponent Polymeric Systems*, CRC Press, 2019.

46 D. Fabbri, A. G. Rombolà, I. Vassura, C. Torri, S. Franzellitti, M. Capolupo and E. Fabbri, *J. Anal. Appl. Pyrolysis*, 2020, **149**, 104836.

47 Y. Zheng, L. Tao, X. Yang, Y. Huang, C. Liu and Z. Zheng, *J. Anal. Appl. Pyrolysis*, 2018, **133**, 185–197.

48 L. Hermabessiere, C. Himber, B. Boricaud, M. Kazour, R. Amara, A.-L. Cassone, M. Laurentie, I. Paul-Pont, P. Soudant, A. Dehaut and G. Duflos, *Anal. Bioanal. Chem.*, 2018, **410**, 6663–6676.

49 M. Fischer and B. M. Scholz-Böttcher, *Environ. Sci. Technol.*, 2017, **51**, 5052–5060.

50 M. Funck, A. Yildirim, C. Nickel, J. Schram, T. C. Schmidt and J. Tuerk, *MethodsX*, 2020, **7**, 100778.

51 T. Ishimura, I. Iwai, K. Matsui, M. Mattonai, A. Watanabe, W. Robberson, A.-M. Cook, H. L. Allen, W. Pipkin, N. Teramae, H. Ohtani and C. Watanabe, *J. Anal. Appl. Pyrolysis*, 2021, **157**, 105188.

52 Y. Liu, R. Li, J. Yu, F. Ni, Y. Sheng, A. Scircle, J. V. Cizdziel and Y. Zhou, *Environ. Pollut.*, 2021, **272**, 115946.

53 M. Kittner, P. Eisentraut, D. Dittmann and U. Braun, *Appl. Res.*, 2024, **3**, e202200089.

54 D. Sorolla-Rosario, J. Llorca-Porcel, M. Pérez-Martínez, D. Lozano-Castelló and A. Bueno-López, *Talanta*, 2023, **253**, 123829.

55 W. Cowger, A. Gray, S. H. Christiansen, H. DeFrond, A. D. Deshpande, L. Hemabessiere, E. Lee, L. Mill, K. Munno and B. E. Ossmann, *Appl. Spectrosc.*, 2020, **74**, 989–1010.

56 R. Peñalver, N. Arroyo-Manzanares, I. López-García and M. Hernández-Córdoba, *Chemosphere*, 2020, **242**, 125170.

57 S. Phan and C. K. Luscombe, *J. Appl. Phys.*, 2023, **133**, 020701.

58 H. Luo, Y. Xiang, Y. Zhao, Y. Li and X. Pan, *Sci. Total Environ.*, 2020, **744**, 140944.

59 A. Popenda, E. Wiśniowska and C. Manuel, *Desalin. Water Treat.*, 2024, **319**, 100456.

60 R. Thavasimuthu, P. Vidhya, S. Sridhar, S. Sasirekha and P. Sherubha, *Polym. Adv. Technol.*, 2024, **35**, e6506.

61 M. P. Kannankai, A. J. Babu, A. Radhakrishnan, R. K. Alex, A. Borah and S. P. Devipriya, *J. Hazard. Mater.*, 2022, **440**, 129801.

62 J.-H. Lee, M.-J. Kim, C.-S. Kim, S.-J. Cheon, K.-I. Choi, J. Kim, J. Jung, J.-K. Yoon, S.-H. Lee and D.-H. Jeong, *Environ. Pollut.*, 2023, **333**, 122017.

63 A. Tirkey and L. S. B. Upadhyay, *Int. J. Environ. Sci. Technol.*, 2024, **21**, 2357–2366.

64 M. Zandieh, X. Luo, Y. Zhao, C. Feng and J. Liu, *Angew. Chem., Int. Ed.*, 2025, **64**, e202421438.

65 B. V. Kumar, L. A. Löschel, H. K. Imhof, M. G. Löder and C. Laforsch, *Environ. Pollut.*, 2021, **269**, 116147.

66 C. Smith, S. Brown, N. Malone, S. Bevers, J. Ranville and D. H. Fairbrother, *Environ. Sci.: Nano*, 2024, **11**, 911–923.

67 J. Ding, C. Sun, C. He, J. Li, P. Ju and F. Li, *Sci. Total Environ.*, 2021, **782**, 146830.

68 N. C. Quaglia, F. Capuozzo, E. Ceci, S. Cometa, A. Di Pinto, A. Mottola, R. Piredda and A. Dambrosio, *Ital. J. Food Saf.*, 2023, **12**, 10906.

69 H.-X. Li, M. Shi, F. Tian, L. Lin, S. Liu, R. Hou, J.-P. Peng and X.-R. Xu, *Sci. Total Environ.*, 2022, **841**, 156749.

70 N. Chinfak, C. Charoenpong, A. Sampanporn, C. Wongpa and P. Sompongchaiyakul, *Mar. Pollut. Bull.*, 2024, **208**, 116937.

71 G. C. Ghosh, S. M. Akter, R. M. Islam, A. Habib, T. K. Chakraborty, S. Zaman, A. H. M. E. Kabir, O. V. Shipin and M. A. Wahid, *Reg. Stud. Mar. Sci.*, 2021, **44**, 101728.

72 S. Gao, K. Yan, B. Liang, R. Shu, N. Wang and S. Zhang, *Sci. Total Environ.*, 2023, **854**, 158575.

73 Y. Tepe, H. Aydın, F. Ustaoğlu and M. Kodat, *Environ. Sci. Pollut. Res.*, 2024, **31**, 55336–55345.

74 S. O. Ogunola, P. Reis-Santos, N. Wootton and B. M. Gillanders, *Mar. Pollut. Bull.*, 2022, **179**, 113706.

75 J. Hara, J. Frias and R. Nash, *Mar. Pollut. Bull.*, 2020, **152**, 110905.

76 M. Rasta, A. Khodadoust, M. S. Taleshi, N. S. Lashkaryan and X. Shi, *Environ. Pollut.*, 2024, **362**, 124959.

77 X. Xiao, S. Liu, L. Li, R. Li, X. Zhao, N. Yin, X. She, W. Peijnenburg, X. Cui and Y. Luo, *Food Chem.*, 2024, **450**, 139317.

78 L. Stabili, E. Quarta and L. Giotta, *Sci. Total Environ.*, 2024, **954**, 176262.