

See discussions, stats, and author profiles for this publication at: <https://www.researchgate.net/publication/323680142>

Microplastics in a Marine Environment: Review of Methods for Sampling, Processing, and Analyzing Microplastics in Water, Bottom Sediments, and Coastal Deposits

Article in *Oceanology* · January 2018

DOI: 10.1134/S0001437017060169

CITATIONS

5

READS

759

2 authors:



Mikhail Borisovich Zobkov

Karelian Research Centre of the Russian Academy of Sciences

25 PUBLICATIONS 182 CITATIONS

[SEE PROFILE](#)



Elena Esiukova

P.P. Shirshov Institute of Oceanology

42 PUBLICATIONS 243 CITATIONS

[SEE PROFILE](#)

Some of the authors of this publication are also working on these related projects:



Propagation of micronized anthropogenic polymer particles (microplastics) and associated heavy metals in large freshwater bodies (Onega Lake as an example) RSF #19-17-00035 [View project](#)



MicroplAstics Research in the BaLtic marine Environment (MARBLE) [View project](#)

INSTRUMENTS
AND METHODS

Microplastics in a Marine Environment: Review of Methods for Sampling, Processing, and Analyzing Microplastics in Water, Bottom Sediments, and Coastal Deposits

M. B. Zobkov* and E. E. Esiukova

Shirshov Institute of Oceanology, Russian Academy of Sciences, Moscow, Russia

*e-mail: duet@onego.ru

Received December 15, 2015; accepted July 3, 2016

Abstract—The basic approaches, methods, and procedures for collecting and analyzing samples of microplastics in a marine environment are briefly described.

DOI: 10.1134/S0001437017060169

Contamination of a marine environment by microplastics is currently an urgent ecological problem barely covered in the Russian scientific press. The main difficulty when providing quantitative assessment of microplastics in marine environment is the absence of standard procedures for collecting and analyzing samples of water, bottom sediments, and coastal deposits.

Today, plastics are one of the most demanded materials used worldwide. The physicochemical properties of plastics, mainly endurance, light weight, and durability combined with low manufacturing costs make this material nearly irreplaceable in the production of domestic goods, construction, and industry.

According to various estimates, the worldwide annual production of plastics ranges from 275 to 299 mln t [13, 26, 29, 37], whereas the scales of utilization and reprocessing are much lower.

Plastic products gradually break down under natural conditions. This results in huge amounts of macro-, micro-, and nanoparticles, which are the most harmful to the environment. The specific density of plastic is close to that of water. Because of this, synthetic litter is easily transported from a catchment area into lakes and rivers and finally enters the seas and the World Ocean [5, 37, 45]. Microplastics have various sizes and low density. As a consequence, many living organisms perceive them as food [2, 4, 19]. Since their enzymatic system cannot break plastic down, ingestion of the latter is harmful by itself for organisms and may cause a fatal outcome [9, 14, 16, 35]. However, the largest concerns arise from the fact that microplastics are able to adsorb contaminants on their surfaces [3, 12, 17, 27, 38] and thereby become a secondary source of contaminants. The latter travel up the food chain and accumulate in higher predators and humans [25, 32].

In addition to the secondary microplastics, which originate from breakdown of staff or large litter debris, there are primary ones that penetrate the water bodies in their initial state [18, 25]. These are plastic granules, or pellets. They serve as a raw material for manufacturing plastic sheets or ready-to-use items. The cosmetic industry [15, 18, 44] uses micro granules (microspheres, nanospheres, microcapsules, nanocapsules) [28, 44]. Currently, there is no final opinion on particle size that corresponds to microplastics, but the majority of researchers [20, 45] agree that these are particles from 0.5 to 5 mm in their larger dimension. Some authors proposed a lower limit close to 0.3 mm [10] due to widespread water sampling techniques using zooplankton nets with a mesh size of 333 μm [18]; the maximum particle size corresponding to the definition of microplastic continues to be a matter of discussion [32]. It is no accident that particles from 0.5 to 5 mm are considered as a special group. This is due to the substantial technical difficulties inherent to analyzing particles smaller than 0.5 mm [20]. In the present review, we adhere to exactly this dimensional range when determining microplastic particles. As yet, there is no clear answer to the question which synthetic substances can be classified as microplastics. This problem has been solved for the majority of polymers, such as polyethylene, polypropylene, polystyrene, etc. However, no solution has been found for other anthropogenic substances such as alkyd resins or viscose.

The first reports on the discovery of microplastics in plankton samples go back to early 1970s [6, 7], but only now is the problem of microplastics in the World Ocean becoming a matter of discussion in foreign scientific literature [5, 11, 15, 25, 40]. Microplastics is an extremely heterogeneous ensemble of particles that vary in size, shape, color, density considerably and may incorporate

a wide variety of synthetic polymers. This is why reliable methods of sample collecting, sample preparation, and plastic detection remain one of the main difficulties in quantitative assessment of the environment. Nearly all researchers raise the question of developing standard procedures. Despite the fact that the first steps in this direction have already been taken [33], the problem as a whole remains unsolved.

Nevertheless, many scientists devote efforts to develop and test techniques for collecting and preparing samples of water, bottom sediments, and coastal deposits, as well as to develop methods for detecting microplastics in samples. Current laboratory procedures for analyzing microplastics in a marine environment developed by NOAA [33] for studying marine litter have certain drawbacks and are voluntary in nature, while monitoring programs require relying on specific solutions depending on the problems to be solved.

To date, the occurrence and detection of microplastics in a marine environment have not been raised in Russian scientific publications. To expand the audience interested in contamination of a marine environment by microplastics and for convenience in searching for relevant information, we have prepared translations of a number of foreign scientific papers on the acquisition and analysis of samples of microplastics in a marine environment, as well as descriptions of procedures and standards for testing the polymers. These translations are available at site of the Shirshov Institute of Oceanology, Russian Academy of Sciences (<http://ioran.ocean.ru/index.php/news/anonsy/item/337-anons-publikatsii>) and involve complete translation of laboratory techniques for analyzing microplastics in a marine environment [33] supplemented by critical comments by authors, discussion of methods for measuring the specific density of noncellular plastic [22, 34], a method for determining plastic in water samples with our improvements, and a complete bibliography of the sources found. Our intention is to expand this list as new reviews appear. Among others, we suggest adding author methods for obtaining and processing samples of microplastics in water, bottom sediments, and coastal deposits based on our own and advanced foreign experiences.

Below is a brief description of the main approaches, methods, and techniques for obtaining and analyzing samples of microplastics in a marine environment.

Different types of equipment [42, 34] and analytical laboratory methods [20, 39] are needed for sampling and determining plastic particles of different size groups. Particle size determines the influence on the life of oceanic communities and migration pathways in the ocean.

Samples of microplastics in a marine environment can be collected (1) from the surface layer, (2) from the water column, and (3) by sampling bottom sediments or (4) coastal deposits. Samples can be (1) selective, (2) bulk, or (3) volume-reduced.

Selective sampling in situ means that plastic debris usually is recognized by the naked eye and is picked up from the sandy beach surface. This technique is useful during assessment of plastic granules and is suitable due to their spherical shape and relatively large sizes (up to several millimeters), which facilitates their recognition on the sandy surface. However, chances of missing plastic are high when it is mixed with other rubbish or has an irregular shape.

No concentration of plastic takes place when **bulk** samples are collected. Such a technique is the most popular when studying bottom sediments, but it is rarely used for water sampling due to low plastic content in water. Bulk sampling is preferable when visual identification of plastic is impossible, i.e., if particles are mixed with the bottom sediments or particles are too small for filtration at the sampling site or for the naked eye recognition.

The **volume-reduced** method is the most applicable for water sampling and sometimes for bottom sediments. In this case, the volume of the initial sample is decreased and only a small fraction of it containing microplastics remains for subsequent analysis. Samples of bottom sediments and sand can be sieved directly at the sampling site on the beach or aboard a vessel, while water samples are usually concentrated by filtration of large volumes of water through plankton nets. The bulk and volume-reduced samples require extra processing under laboratory conditions.

According to the NOAA recommendations [33], the following procedures can be used for separating microplastic particles from environmental samples depending on their size: (1) flow Nanofiltration or nanofractionation of particles smaller than 1 μm ; (2) a standard filter for particles sizing from 0.2 to 1 μm ; (3) the use of phytoplankton neuston net 50–80 μm (*Phyto-P net*); (4) zooplankton neuston net (*Zoo-P net*) 330 μm ; (5) a 5 mm sieve for sieving. It is difficult to compare the results from sieving and plankton nets. Therefore, it is recommended to use sieves with a 500 μm mesh and separately process the particles retaining and passing through the sieve as a mandatory operation within each of the above procedures [20]. This makes it possible to compare the results from density separation and filtering to the data obtained from analysis of larger fractions (0.5–5 mm) with sieves.

Surface water samples. Neuston nets are mainly used in this case. The main advantage is fast filtering of large volumes of water and obtaining of a concentrated sample. The nets make it possible to get a representative sampling from a large water surface, to collect plastic particles of the millimeter range, and concurrently collect microplastics and zooplankton to compare their amounts.

The mesh and opening sizes are the most important characteristics of sampling nets. However, the parameters of sampling nets are rarely reported in full

and the mesh size is usually the only parameter mentioned in publications. The mesh size depends on the research goals and varies from tens of microns to millimeters, but it corresponds to the mesh size of zooplankton nets on average. The opening size of neuston nets is up to 2 m². The length of these nets for surface sampling varies substantially from one to several meters, but nets 3–4 m long are the most common. The surface layer sampled by such nets is about 15–25 cm thick. The buoyancy of a net is supported by special floats (*manta trawl*) or catamaran (*neuston catamaran*). The net towing speed varies from 1 to 5 knots. The use of *catamaran* facilitates sampling in rough sea conditions while the efficiency a *manta trawl* decreases due to submersion of its inlet in waves. Therefore, the latter net is applicable in calm waters. Duration of towing varies from minutes to hours depending on the SPM content in water.

The use of nets with different mesh sizes considerably hampers comparison of the measurement results.

Samples from the water column. Zooplankton nets, a continuous plankton recorder (CPR) [43], a near-bottom trawl for catching benthic organisms (*epibenthic sled*) [31], and various submersible pumps are the most commonly used tools in the water column. The water intake system of a research vessel is used too, but rarely. Niskin bottles are employed for full-volume sampling of waters. Different studies used different methods for water sampling at depth ranges from one to hundreds of meters.

Samples of coastal deposits. The samples were mainly collected at different parts of a beach and the choice of the specific site can influence the analysis results. The sample collection (or series of collections) is performed (1) over the entire beach, (2) within a number of separate zones, (3) along the alignment of different zones of a beach, (4) along the line of maximum tide or uprush (on the upper beach), and (5) in ditches or trenches beyond the beach.

Simple tools are frequently used for collecting samples of plastic granules and fragments (tweezers, metal spoons, or scoops) or particles are simply collected by hand using a special container or bag. The collecting strategy varies: movement in the same direction along the shoreline and picking up material with a spoon or scoop or sometimes from a certain area within a special frame. In a number of cases, stratified sampling with special tubes is used.

Sampling units directly depend on the tools used. In the case of frames or pipes, the concentrations were calculated per area (from several cm² to 5 m²). In other cases, the concentration is calculated per sample weight, which can range from hundreds of grams to 10 kg, or per sample volume, which varies from tens of milliliters to several liters.

As a rule, the sampling comprises the upper 5 cm of sediment, but it may be limited to the surface of a beach or as deep as tens of centimeters. Microplastics

are able to accumulate in sand in the same way as organic particles and can be buried in deeper layers. In this case, it is desirable to use a stratified sampling procedure with a tube.

Bottom sediments samples. Procedures for bottom deposits sampling for microplastics content are similar to those to determine their chemical composition or for biological assessment. The samples are collected by grabs (Ekman-Birdge, Van-Veen, Peterson), a bottom trawl, or core sampler. The distribution of microplastics as particulate matter in bottom sediments may be substantially heterogeneous. Therefore, it is necessary to perform several sediment samplings to obtain a single representative sample when using point-by-point devices such as a grab or a corer. This is particularly important in the case of compact samplers.

Conservation, storage, and quality control. Incorrect storage of samples can cause a change in their particle size distribution due to partial destruction of plastic particles and, as a consequence, the impossibility of their detection by available methods. Therefore, when storing or transporting samples, it is desirable to use procedures that can maintain the plastic in its initial state at the moment of sampling from the environment.

The principles of storage and conservation of samples before laboratory analysis: (1) in darkness (if possible, at 4°C regardless of sample matrix), (2) freezing (down to –20°C) the samples of bottom sediments and filters, (3) the use of various fixing solutions (ethanol, DESS, 4–5% formalin solution, 5–10% HCl solution), (4) combined use of fixing solution and storage at 4°C, (5) drying at room temperature and storage in the dark (shipborne analysis).

It is proper to avoid and replace when possible the potential sources of plastic contamination (plastic laboratory ware, plastic sieves and devices) for metal or glassware. However, it is impossible in the case of nets and a number of other instruments. A control with empty samples is obligatory when plasticware is used for storing the samples. The contamination of samples may be due to insufficient air quality in the laboratory, particles of synthetic working cloths, poor cleaning of tools, loosely closed containers with samples, paint particles broken off from the side of the vessel by samplers, or particles of synthetic nets used for water sampling. Blank samples must be used in routine for contamination monitoring.

Sample preparation. There are four main stages: (1) density separation, (2) filtering, (3) sieving, and (4) organic matter digestion. All are aimed at separating microplastic particles from the main sample material (water, bottom sediments, and sand) and removing of organic material.

Density separation. The specific density of most plastics ranges from 0.8 to 1.70 g/cm³. The range extends down from <0.05 g/cm³ for expanded polystyrene and up to 2.1–2.3 g/cm³ for polytetrafluoroethylene/Teflon [8]. Generally, the density of sand

and other deposits is 2.65 g/cm^3 . This difference is used widely to separate comparatively light plastics from heavier soil particles by placing the sample in a saturated saline solution and mixing it for a certain period of time. After mixing, sand and bottom sediments settle out, while lighter particles (including plastic ones) remain suspended or float on the solution surface. The floating particles are collected for further processing. As a separating solution, fresh or tap water, sea water, a concentrated NaCl solution (with a density of 1.2 g/cm^3), sodium polytungstate, lithium metatungstate, zinc chloride, sodium iodide, and other salts are used. Lightweight and foamed plastics, whose density is less than 1 g/cm^3 , can also be separated using fresh water. Plastics that float in seawater include expanded polystyrene and high- and low-density polyethylene and polypropylene. Polystyrene in solid form emerges only in a saturated saline NaCl solution. Elastic and rigid polyvinyl chloride (PVC), polyethylene terephthalate (PET), and nylon float up in a sodium metatungstate solution. Since the density of some plastics reaches 1.7 g/cm^3 , the use of a saturated solution of sodium chloride and especially fresh water can lead to underestimation of the total microplastic content. Various instruments (shakers, mixers, centrifuges, flotators, and separators) are employed to mix the solution. In the simplest case, the sample is placed in a beaker and mixed with a glass stir rod. The time of mixing is one of the main parameters of the extraction. It can vary considerably depending on the volume of the sample and can range from tens of seconds to several hours. The settling time also varies greatly and ranges from a few minutes to a day. It is also expedient to carry out sequential extractions from the sediment, which leads to increase in the efficiency of microplastics extraction significantly, but at the expense of increasing the analysis time also. The most effective extraction method at this time involves the Munich Plastic Sediment Separator (MPSS) [21].

Filtration. Vacuum filtration is usually employed for a solution obtained at the density separation stage that contains afloat plastic particles. For this, fiberglass, polycarbonate membrane, paper, nitrocellulose, and silicon filters specially designed for FT-IR spectroscopy are used. The pore size varies from 1 to $1.6 \mu\text{m}$. Filters with a pore size of $2 \mu\text{m}$ are rarely used. Sometimes, the working solution with afloat particles is passed through a fine mesh.

Filtration is also used to isolate microplastics from bulk water samples. In this case, filters up to 15 cm in diameter and with a pore size of up to $47 \mu\text{m}$ are used.

It is possible to collect microplastic particles from the surface of the solution with tweezers. For separation of large particles, samples before filtration can be passed through a sieve with a mesh size of $500 \mu\text{m}$ [20]. An important point is the process of moving the floating particles from the surface of the solution to the filters and sieves. To prevent any loss of analyte associ-

ated with the adherence of particles to the walls of the laboratory ware, it is recommended to sequentially wash the walls of glassware directly on the filter.

Sieving. Microplastics can be isolated from samples by sieving the latter through sieves with different mesh sizes. The material on the sieves is further sorted, and the remainder that has passed through the sieve is discarded. The use of sieves with meshes of various sizes makes it possible to separate microplastic particles into several size groups. A cascade of several sieves (one to six) with sizes of 0.038 to 4.75 mm is usually used. The sieve material is usually stainless steel or copper.

Filters and sieved material is dried at room temperature or in desiccators. The temperature in the desiccators varies greatly in different methods (from 60 to 90°C), but the standard [24] sets the conditions for preparing plastic samples before testing and recommends not to exceed the temperature of $50 \pm 2^\circ\text{C}$, with a drying time of 24 h with subsequent bringing the temperature to normal conditions in a desiccator. This is established to prevent changes in the composition and physicochemical properties of the plastics.

Organic matter digestion. The amount of natural organic matter (algae, zooplankton, and phytoplankton and remnants of tests and shells of marine organisms) in samples can significantly exceed the volume of the analyte. Plastic particles are susceptible to fouling by some forms of brown algae and bacterial film. This can introduce an error in determining certain physical characteristics, e.g., specific density. To eliminate the effect of biomaterial dissolution in alkalis, acids, and oxidizers or decomposition by enzymes are applied. Sometimes ultrasonic washing of plastic particles in distilled or deionized water is used to remove possible surface contamination by sand or silt.

It is important to use proper identification techniques for analysis of different size groups of plastics. Today, visual detection methods, pyrolysis gas chromatography with mass spectrometry, etc. are widely used, but the spectrometric techniques yield the most qualitative identification results.

Identification of microplastics. Visual examination (with the naked eye or a microscope) and sorting of concentrated samples are often the first step in separating plastic from organic residues and other non-plastic waste, such as glass or resin. In the case of visual examination without a microscope, attribution of a particle to the plastics is usually determined by such subjective characteristics as gloss, brightness, unusual color, shape, structure or elasticity or hardness determined by tweezers.

Microplastic particles larger than 1 mm can be visually detected under a microscope in accordance with the following rules [36]:

- (1) Cell structure and other organic forms of particles are absent.
- (2) Fibers should have uniform color and thickness along the entire length.

(3) Particles must have a clean and uniform color.

(4) If they are transparent or a white, then they should be examined by a fluorescence microscope at large magnification.

In some cases, it is proposed that white, transparent, and black particles should not be accounted as plastics, because they interfere with biological material and other substances. This approach deliberately underestimates the microplastic content and should be applied with caution.

It is expedient to use pyrolysis gas chromatography with mass spectrometry (pyrolysis GC-MS, PyrGC-MS) and FT-IR spectrometry.

Visual methods should not be used separately from chemical or spectroscopic ones (the latter should be used to confirm the results of visual determination). To identify microplastics of lower size groups, only the spectrometric method should be used (although this method is very time-consuming) [41]. The choice of a representative detection method is of paramount importance in evaluating microplastics contamination.

Microplastic particles found in a marine environment can be characterized by several criteria, such as size, shape, specific density, color, chemical composition, and concentration in water. Therefore, microplastics analysis can be divided into two main components: morphological description with determination of the physical and chemical characteristics of particles and quantitative analysis with determination of the chemical composition of the polymers.

Physicochemical characteristics. Particle size is the main characteristic, with the exception of chemical composition. The size of a particle is usually understood as its length according to the largest dimension. The size can be determined directly, using measuring instruments, e.g., a microscope with a graduated scale, a digital microscope, or a caliper in visual analysis; indirectly using a set of sieves with separation of a sample of microplastics into size groups; or with combined use of sieves and measuring instruments. The size groups of particles assigned during the analysis depend directly on the sampling and separation techniques used and are related to the nominal sizes of the filters or sieves involved.

Microplastics particles differ in shape: they can be spherical, irregular, or long fibers. The shape of particles depends on the degree of destruction and the residence time in the environment. For morphological characterization of the surface of plastics, scanning electron microscopy (SEM) and some of its variants are used, but no method has yet been developed to determine how long the particle had been in the marine environment.

The number of categories used to classify microplastic particles depends on the aim of the study and varies greatly. The morphological description of microplastics is based on the origin, type, shape, color, and/or degree of degradation. The reference collections describing the

characteristics of the particles found in the sea have high significance.

Color is one of the main characteristics of plastics when using visual identification. The most common color is white or close to it (faded yellow, cream-colored). The color of particles can introduce uncertainty into the extraction process in cases where plastic is hidden by a large number of biological residues. Particles with bright colors are highly likely to be selected as plastic, while those with faded colors can easily be missed, thus contributing a fraction of bias to the analysis process. Color is used to assess the degree of photodegradation, the residence time of a particle on the water surface, and the degree of fouling and weathering.

A number of indirect characteristics, such as specific density or color, make it possible to roughly identify the polymer of which the particle consists. However, to make a final decision, chemical analysis is needed.

For a rough identification of the polymer type, subjective characteristics such as the color of the flame or smell of smoke released during its burning can be used.

Although plastics belong to amorphous materials, many of them (HDPE, PVD, PP, PET, etc.) have a partial crystal structure and can be characterized by a certain melting point, which can be used for an approximate evaluation of the type of polymer. The melting point of plastic granules can be evaluated by differential scanning calorimetry.

To determine the specific density, it is expedient to use the method proposed by Kolb and Kolb [30], the method for measuring the density of noncellular plastics by the titrimetric method [22], or a similar flotation method [1]. However, it is worth considering that these methods are designed to determine the density of noncellular plastics only.

The use of such indirect characteristics as density, color, or melting point can be useful for rapid and inexpensive determination of the polymer type of plastic granules, since the granules manufacturer describes these characteristics. However, this approach cannot be used for plastic fragments, since their shape and color vary within a wider range, in particular, due to fragmentation and erosion, and cannot be unambiguously associated with a particular type of plastic.

Determination of the chemical composition of microplastics and the type of polymer. To identify microplastics, analytical techniques such as pyrolysis GC-MS, Raman spectroscopy, SEM, and some of its varieties are used, as well as IR Fourier spectrometry.

Pyrolysis GC-MS is used to assess the chemical composition of microplastic particles by analyzing the gaseous products of their thermal decomposition. A polymer is identified by characteristic pyrograms, via their comparison with reference pyrograms of known pure polymer samples. Pyrolysis GC-MS makes it possible to determine the types of polymers with sufficient accuracy. However, in the case of analysis of microplastics, this approach has a significant disad-

vantage: particles must be manually placed in a pyrolysis tube. Since manual manipulation is applicable exclusively to relatively large particles, this imposes a limit on the minimum size of microplastics to be analyzed. In addition, the method allows one to analyze only one particle per cycle, and is therefore poorly suited for processing a large number of samples.

Raman spectroscopy is an efficient chemical analysis method. In contrast to IR-spectrometry, Raman scattering results from inelastic scattering of light by molecules of a substance, and scattered light differs from the exciting one in terms of wavelength. Advantages of this technology are no need for sample preparation, the ability to conduct measurements directly in water, and the use of glass or quartz cells for measuring. By combining this method with microscopic analysis, it is possible to obtain a more powerful tool for the study of microplastics – Raman microspectroscopy, which also provides information on the crystal structure of a polymer. Raman microspectroscopy makes it possible to determine the type of a polymer for ultrasmall plastic particles up to a few microns in size.

At present, IR spectrometry is most commonly used to identify microplastics. This method compares the IR absorption spectrum or transmission of a sample with the spectra of known synthetic substances. For this, various spectrometric instruments are used: an infrared spectrometer, an IR Fourier spectrometer, and an infrared spectrometer for near-IR operation. They can determine a wide range of polymers, e.g., PP, PE, and polyester. However, for microplastic particles studied on a conventional IR spectrometer, technical difficulties arise due to the small particle size. This problem can be solved by using micro-IR Fourier spectrometry, as well as measurement of spectra under conditions of attenuated total reflectance (ATR). In some cases, these approaches can be achieved by purchasing additional equipment: a special microscope or ATR-FTIR cells available for many modern FTIR spectrometers. The combination of these two approaches (micro-ATR and FT-IR analyses), in addition to measuring the particle spectra, makes it possible to add visualization and mapping of samples and to automate the determination of the polymer type. However, this procedure is very expensive, time-consuming, and requires highly qualified specialists; however, it is most suitable for identifying microplastic particles and exhibits high reliability and reproducibility. In connection with the laboriousness of spectrometric analyzers, they are often used to determine not all the material obtained from the sample, but only some fraction of the latter.

To increase the effectiveness of monitoring programs, it should be remembered that the choice of an appropriate method for selecting and identifying microplastics is crucial for assessing this type of contamination [41]. When planning monitoring studies, it is important, if possible, to adhere to the most strin-

gent requirements for sampling, preparation, and analysis of samples while retaining the maximum amount of information for comparison of the results obtained by different research groups in order to estimate the magnitude of the problem on a global scale. When developing new methodological approaches, it should be remembered that plastics are industrial materials whose testing methods are already widely known. These methods, with some modifications, can be improved and implemented for analysis of microplastics. Some standards [23] describe sample preparation techniques that can also be adapted for use in this field.

We hope that this material will help other researchers in solving problems of estimating the amount and dynamics of microplastics in a marine environment and determining its role in the world pollution.

ACKNOWLEDGMENTS

This work was supported by the Russian Science Foundation, project no. 15-17-10020.

REFERENCES

1. *GOST (State Standard) 15139-69: Plastics. Methods for the Determination of Density (Mass Density)* (Izd. Standartov, Moscow, 1981) [in Russian].
2. A. B. Aloy, B. M. Vallejo, Jr., and M. A. Juinio-Meñez, "Increased plastic litter cover affects the foraging activity of the sandy intertidal gastropod *Nassarius pullus*," *Mar. Pollut. Bull.* **62**, 1772–1779 (2011).
3. K. Ashton, L. Holmes, and A. Turner, "Association of metals with plastic production pellets in the marine environment," *Mar. Pollut. Bull.* **60**, 2050–2055 (2010).
4. C. M. Boerger, G. L. Lattin, S. L. Moore, and C. J. Moore, "Plastic ingestion by planktivorous fishes in the North Pacific Central Gyre," *Mar. Pollut. Bull.* **60**, 2275–2278 (2010).
5. M. A. Browne, P. Crump, S. J. Niven, et al., "Accumulations of microplastic on shorelines worldwide: sources and sinks," *Environ. Sci. Technol.* **45**, 9175–9179 (2011).
6. E. J. Carpenter, S. J. Anderson, G. R. Harvey, et al., "Polystyrene spherules in coastal waters," *Science* **17**, 749–750 (1972).
7. E. J. Carpenter and K. L. Smith, "Plastics on the Sargasso Sea surface," *Science* **175**, 1240–1241 (1972).
8. I. Chubarenko, A. Bagaiev, M. Zobkov, and E. Esiukova, "On some physical and dynamical properties of microplastic particles in marine environment," *Mar. Pollut. Bull.* **108**, 105–112 (2016).
9. M. Cole, H. Webb, P. K. Lindeque, et al., "Isolation of microplastics in biota-rich seawater samples and marine organisms," *Sci. Rep.* **4** (4528), (2014). doi: 10.1038/srep04528
10. A. Collignon, J.-H. Hecq, F. Glagani, et al., "Neustonic microplastic and zooplankton in the North Western Mediterranean Sea," *Mar. Pollut. Bull.* **64**, 861–864 (2012).
11. M. H. Depledge, F. Galgani, C. Panti, et al., "Plastic litter in the sea," *Mar. Environ. Res.* **92**, 279–281 (2013).

12. S. Endo, R. Takizawa, K. Okuda, et al., "Concentration of polychlorinated biphenyls (PCBs) in beached resin pellets: variability among individual particles and regional differences," *Mar. Pollut. Bull.* **50**, 1103–1114 (2005).
13. M. Eriksen, N. Maximenko, M. Thiel, et al., "Plastic pollution in the South Pacific subtropical gyre," *Mar. Pollut. Bull.* **68** (1–2), 71–76 (2013).
14. P. Farrell and K. Nelson, "Trophic level transfer of microplastic: *Mytilusedulis* (L.) to *Carcinusmaenas* (L.)," *Environ. Pollut.* **177**, 1–3 (2013).
15. L. S. Fendall and M. A. Sewell, "Contributing to marine pollution by washing your face: microplastic in facial cleansers," *Mar. Pollut. Bull.* **58**, 1255–1228 (2009).
16. E. M. Foekema, C. De Gruijter, M. T. Mergia, et al., "Plastic in North Sea fish," *Environ. Sci. Technol.* **47** (15), 8818–8824 (2013).
17. J. P. G. L. Frias, P. Sobral, and A. M. Ferreira, "Organic pollutants in microplastics from two beaches of the Portuguese coast," *Mar. Pollut. Bull.* **60**, 1988–1992 (2010).
18. *Sources, Fate and Effects of Microplastics in the Marine Environment: A Global Assessment*, Ed. by P. J. Kershaw (International Maritime Organization, London, 2015), No. 90.
19. E. Graham and J. Thompson, "Deposit- and suspension-feeding sea cucumbers (Echinodermata) ingest plastic fragments," *J. Exp. Mar. Biol. Ecol.* **368**, 22–29 (2009).
20. V. Hidalgo-Ruz, L. Gutow, R. C. Thompson, and M. Thiel, "Microplastics in the marine environment: a review of the methods used for identification and quantification," *Environ. Sci. Technol.* **46**, 3060–3075 (2012).
21. H. K. Imhof, J. Schmid, R. Niessner, et al., "A novel, highly efficient method for the separation and quantification of plastic particles in sediments of aquatic environments," *Limnol. Oceanogr.: Methods* **10**, 524–537 (2012).
22. *ISO 1183–1:2012: Plastics—Methods for Determining the Density of Non-Cellular Plastics. Part 1: Immersion Method, Liquid Pycnometer Method and Titration Method* (International Organization for Standardization, Geneva, 2012)
23. *ISO 4611:1999: Plastics—Determination of the Effects of Exposure to Damp Heat, Water Spray and Salt Mist* (Association Française de Normalisation, La Plaine Saint-Denis, 1999).
24. *ISO 4611:1987: Plastics—Determination of the Effects of Exposure to Damp Heat, Water Spray and Salt Mist* (International Organization for Standardization, Geneva, 1987).
25. J. A. Ivar do Sul and M. F. Costa, "The present and future of microplastic pollution in the marine environment," *Environ. Pollut.* **185**, 352–364 (2014).
26. J. R. Jambeck, R. Geyer, C. Wilcox, et al., "Plastic waste inputs from land into the ocean," *Science* **347** (6223), 768–771 (2015). doi 10.1126/science.1260352
27. H. K. Karapanagioti and I. Klontza, "Testing phenanthrene distribution properties of virgin plastic pellets and plastic eroded pellets found on Lesvos Island beaches (Greece)," *Mar. Environ. Res.* **65**, 283–290 (2008).
28. *Sources, Fate & Effects of Micro-Plastics in the Marine Environment—A Global Assessment: Report of the Inception Meeting, March 13–15, 2012*, Ed. by P. J. Kershaw and H. Leslie (UNESCO, Paris, 2012).
29. A. A. Koelmans, "ET&C perspectives," *Environ. Toxicol. Chem.* **33**, 5–10 (2014).
30. K. E. Kolb and D. K. Kolb, "Method for separating or identifying plastics," *J. Chem. Educ.* **68**, 348 (1991).
31. G. L. Lattin, C. J. Moore, A. F. Zellers, et al., "A comparison of neustonic plastic and zooplankton at different depths near the southern California shore," *Mar. Pollut. Bull.* **49**, 291–294 (2004).
32. *Marine Anthropogenic Litter*, Ed. by M. Bergmann, (Springer-Verlag, New York, 2015). ISBN 978-3-319-16509-7. doi 10.1007/978-3-319-16510-3
33. J. Masura, J. Baker, G. Foster, C. Arthur, and C. Herring, *Laboratory Methods for the Analysis of Microplastics in the Marine Environment: Recommendations for Quantifying Synthetic Particles in Waters and Sediments NOAA Technical Memorandum, NOS-OR&R-48* (NMFS Scientific Publications Office, Seattle, WA, 2015).
34. S. Morét-Ferguson, K. L. Law, G. Proskurowski, et al., "The size, mass, and composition of plastic debris in the western North Atlantic Ocean," *Mar. Pollut. Bull.* **60**, 1873–1878 (2010).
35. F. Murray and P. R. Cowie, "Plastic contamination in the decapod crustacean *Nephrops norvegicus* (Linnaeus, 1758)," *Mar. Pollut. Bull.* **62**, 1207–1217 (2011).
36. F. Norén, *Small Plastic Particles in Coastal Swedish Waters*, KIMO Report (Lysekil, 2007).
37. M. C. Rilling, "Microplastic in terrestrial ecosystems and the soil?" *Environ. Sci. Technol.* **46**, 6453–6454 (2012).
38. L. M. Rios, P. R. Jones, C. Moore, and U. V. Narayan, "Quantitation of persistent organic pollutants adsorbed on plastic debris from the Northern Pacific Gyre's "eastern garbage patch," *J. Environ. Monit.* **12**, 2226–2236 (2010).
39. T. Rocha-Santos and A. C. Duarte, "A critical overview of the analytical approaches to the occurrence, the fate and the behavior of microplastics in the environment," *Trends Anal. Chem.* **65**, 47–53 (2015).
40. C. M. Rochman, M. A. Browne, B. S. Halpern, et al., "Policy: classify plastic waste as hazardous," *Nature* **494**, 169–171 (2013).
41. Y. K. Song, S. H. Hong, M. Jang, et al., "A comparison of microscopic and spectroscopic identification methods for analysis of microplastic in environmental samples," *Mar. Pollut. Bull.* **93** (1–2), 202–209 (2015).
42. Y. K. Song, S. H. Hong, M. Jang, et al., "Large accumulation of micro-sized synthetic polymer particles in the sea surface microlayer," *Environ. Sci. Technol.* **48**, 9014–9021 (2014).
43. R. C. Thompson, Y. Olsen, R. P. Mitchell, et al., "Lost at sea: where is all the plastic?" *Science* **304**, 838 (2004).
44. *Plastic in Cosmetics* (United Nations Environment Program, Nairobi, 2015). ISBN: 978-92-807-3466-9.
45. S. L. Wright, R. C. Thompson, and T. S. Galloway, "The physical impacts of microplastics on marine organisms: a review," *Environ. Pollut.* **178**, 483–492 (2013).

Translated by G. Karabashev