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Review

Microplastics in Agricultural Systems: Analytical Methodologies and Effects on Soil Quality and Crop Yield

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Abstract: Around one million metric ton of plastics is produced worldwide daily. Plastic contamination is aggravated when the particles reach sizes between 5 mm and 1 µm, giving rise to microplastics, which are omnipresent in the environment, especially in agroecosystems. To appreciate the magnitude of this problem, this review analyzes 177 scientific works to focus on the occurrence and effects of microplastics in agricultural soils. Firstly, the sources, behavior and fate of microplastics in agroecosystems are evaluated. Then, in the absence of a standard methodology for the study of microplastics in farmland soils, the procedures which have been employed for microplastic separation (density and floatation in 73% of the discussed works), identification and quantification (stereomicroscopy, 77%; infrared analysis, 62%) are addressed to provide a practical work guideline. Finally, we highlight the interaction between microplastics and soil microbiota, fauna and vegetation (negative effects reported in 83% of cases), including crop production (decrease in growth parameters in 63% of the reports). From this review, it can be inferred that microplastics may disrupt the biophysical environment of farmland soil, potentially leading to economic losses and to their entrance into the trophic food chain, affecting human feeding and health.

Keywords: agricultural soil; microplastics; analytical methods; soil physicochemical properties; soil fauna; soil microbiota; crop production

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1. Introduction

Plastic pollution in the environment has reached an unprecedented level [1]. To understand this global problem, it is necessary to analyze human influence from the perspective of the evolution of science and technology. Since the first large-scale manufacture of synthetic plastic (Bakelite) in the 1940s–1950s, the global production of plastics has soared, especially in the last 50 years [2,3]. In fact, 367 million metric tons (Mt) of plastics were produced in 2020 around the world [4]. It has also been estimated that 8078 Mt of plastic waste has been generated between 1950 and 2020 [2,4], and around 4900 Mt of all plastic produced throughout history is accumulating in landfills or in the natural environment [2]. Therefore, plastics are currently ubiquitous in the environment, even being proposed as a geological indicator of the Anthropocene period [5]. The problem worsens when plastic debris reaches a smaller size, whether intentionally during production or through the abiotic and biotic degradation of bigger pieces (e.g., thermo-oxidation, photo-oxidation, atmospheric oxidation, mechanical and microbial activities), since the bioavailability, toxicity

and transport processes of plastic particles in ecosystems are promoted when their size is lower [6–8].

Microplastics (MPs) are plastic particles with a size of less than 5 mm and down to 1 μm, or even 0.1 μm, depending on the authors [9,10]. Recently, the terrestrial environment has been recognized as the real source and sink of plastics, especially agricultural soils [10–12]. These findings highlight the need to investigate how MPs can contribute to the stability of agroecosystems, particularly considering that agricultural lands represent almost half of the global land surface, and also that healthy soils are the basis for global food safety and crop production [13]. In this regard, in just the last few years, several authors have provided alarming evidence on the potential impact of MPs on all aspects of agricultural systems [6,9,14–18]. Soil physical properties may be affected by the presence of MPs, yielding changes in soil bulk-density, porosity, aggregate formation, water distribution availability, etc. MPs also impact soil chemistry, disrupting the C, N and P cycles, therefore affecting flora nutrient uptake and food-crop production [14,15,17]. MPs constitute new ecosystems for microbial communities, which show differences regarding those typically found in agricultural soils, and also act as vectors of pathogens and contaminants [16,18]. Soil inhabitants may ingest MPs, suffering injuries and spreading MPs themselves and their co-pollutants [6,9]. Within this context, it is reasonable to seek improvement in the methodologies for MP quantification and characterization in edaphic systems [7,8,19–22]. For these reasons, the study of MPs in agricultural soils is fast advancing, although it is still at an early stage. Therefore, it is essential to update recent findings and put forward future research directions [23].

This review aims to evaluate the existing research on MPs in the agro-environment and to provide an up-to-date overview of the impact of MPs from the point of view of agricultural quality and crop production. Thus, the main objectives can be stated as follows: (i) to show the current state of MP studies in agricultural soils; (ii) to summarize the most common sampling, extraction, quantification and identification methods for MPs in agricultural soils; (iii) to provide an overview of the abundance of MPs in agricultural soils; (iv) to assess the effects of MPs on soil physicochemical properties, fauna, microbiota and vegetation; and (iv) to outline the knowledge gaps that require further investigation regarding the presence of MPs in agricultural soils.

2. Methodology

The consulted literature was found on different scientific search engines (Web of Science, Scopus, ResearchGate and Google Scholar, as well as editorial webpages) using the following combination of key topics: "microplastic in agricultural soil", "extraction methods", "physicochemical properties", "fauna", "microbiota" and "vegetation", among others. Only published, peer-review experimental studies were analyzed in this revision. Experimental studies were mainly analyzed while reviews were consulted for complementary information. In total, 137 research articles were consulted (see Table S1 of Supplementary Material). From the selected literature, 52 papers focused on the assessment of the amounts of MPs and extraction and quantification methods, and 85 on MP transport processes and their effects on soil properties and associated biota; of the latter, 18 were specifically about the impact of MPs on agricultural production (research about hydroponic crops was mainly omitted). The papers were first sorted by their objectives and their analyzed effects, and then, schematically summarized by intersecting topics.

3. Origin and Fate of MPs in Agricultural Soils

3.1. Sources

Agricultural systems are particularly high receptors of MPs, mainly because of soil management techniques and, to a lesser extent, due to atmospheric deposition [24–26]. Numerous agricultural practices carried out for increasing crop yields and maintaining soil moisture, quality and nutritional status may, ironically, affect soil health due to MP accumulation [6,27]. For instance, in conventional farming, a huge number of plastic

products (e.g., mulching, packaging, greenhouse shedding, seedbeds and water pipes) are used, some of which eventually disintegrate on the field and transform into MPs through a combination of physical, chemical and biological effects [7,9,19,22,27–29]. Indeed, plastic mulching has become a major source of MPs since it is often left in agricultural soils to avoid disposal costs [30,31]. As an example, Huang et al. observed how the abundance of MP particles in soils increased after continuous plastic mulching applications [30]. In addition, Yang and coworkers monitored the degradation of typical mulching films, finding MP generation to be between 147 and 475 particles cm^{-2}; biodegradable films fragmented the fastest, followed by oxo-degradable and conventional polyethylene (PE) films [32].

Other common agricultural practices such as irrigation and organic fertilization, with compost obtained from municipal solid wastes, manure or sewage sludge, have been detected as an important entry pathway of MPs into agricultural soils [10,33,34]. Compost produced from household wastes may include both plastic debris from packaging materials and food waste contaminated with MPs, while sewage sludge may contain MPs via laundry, personal care products and urban runoff [35–38]. In this respect, wastewater treatment plants play a significant role, with the removal rates of MPs generally over 95%, depending on the technologies included [39]. Thus, MPs are mainly retained in sewage sludge, while a lesser part remains in the final effluents; both are recycled, in many countries, as soil fertilizer and irrigation water, respectively [27,40–43]. MPs have also been detected in river water for agricultural irrigation [44]. Plastic coatings from slow-release fertilizers and films from coated seeds have also been detected as potential sources of MPs in agricultural soils [45,46]. Until now, there has been no evidence of MP occurrence in inorganic fertilizers [40]. Table 1 shows the characteristics and concentration of MPs, grouped according to the potential source identified in agricultural soils.

Table 1. Characteristics and density of MPs from different sources in agricultural soils.

Potential Source	Study Area (Country)	Depth (cm)	Concentration [a]	Dominant Shape	Dominant Size (mm)	Polymers	Reference
Plastic mulching	Applied over 5, 15 and 24 years (China)	0–40	80.3 ± 49.3 (5 years) 308 ± 138.1 (15 years) 1075.6 ± 346.8 (24 years)	Fragment	<1	PE	[30]
	Applied at least once per year over the last 10 years (Spain)	0–10	2116 ± 1024	(-)	<5	(-)	[47]
	Historically applied (China)	(-)	29.3 ± 33.1 [b]	Fragment (39.6%) Fiber (43.7%) Film (16.1%) Pellet (0.6%)	0–1	PP (27.4%) Rayon (23.5%) PE (18.8%) PET (9.7%) PE (film)	[48]
	Applied over 32 years (China)	0–100	7183–10,586 (0–10 cm) 8885 (80–100 cm)	Film Fiber	(-)	Rayon Polyester Terephthalic acid PP PET (fibers)	[49]
Sewage sludge	30 fields successively treated over 10 years (Chile)	0–25	1.1–3.5 [b]	Fiber (97%)	<2	PES PE PVC Nylon Acrylic	[40]
	Untreated/treated (20–22 t ha^{-1} between 1 and 8 applications) (Spain)	0–30	930 ± 740 light density and 1100 ± 570 heavy density (untreated) 2130 ± 950 light density and 3060 ± 1680 heavy density (treated)	Fragment (80–90%) Film Fiber	0.15–0.25	PP PVC	[50]
	Untreated/treated (three types of sludge: fresh municipal sludge, fresh mixed sludge, and dry heat-treated municipal sludge) (China)	0–20	40.2 ± 15.6 (untreated) 68.6 ± 21.5–149.2 ± 52.5 (treated)	Fiber (66.7–82.5%)	1–3	PP (47.8%) PES (39.1%) PE (6.0%) Rayon (7.1%)	[34]
	Untreated/treated (treated for 5 years *vs.* recently treated) (Spain)	0–15	31–120 (untreated) up to 177–235 (treated 5 years) up to 138–288 (recently treated)	Fiber (44–91%) Fragment (4–44%)	>1 (fiber) <0.5 (fragments)	Acrylic (22–37%) PES (22–53%) PE PP PA	[51]

Table 1. *Cont.*

Potential Source	Study Area (Country)	Depth (cm)	Concentration [a]	Dominant Shape	Dominant Size (mm)	Polymers	Reference
Other organic fertilizers (e.g., biosolids, compost and manure)	Three untreated/treated fields (before and after application of biosolids) (Canada)	0–15	18 ± 22.2%, 187 ± 53.1% and 541 ± 56.4 (before) 25 ± 20.8%, 130 ± 23.1% and 298 ± 39.1% (after)	Fiber (41–45%)	>0.05	PP PE PES Acrylic	[52]
	Untreated/treated (with 22 years of application of pig manure) (China)	~20	16.4 ± 2.7 (untreated) 43.8 ± 16.2 (treated)	Fiber (untreated) Fiber Fragment Film Granule (treated)	<0.5	PES (39.1%) PP (47.8%) Rayon (7.1%) PE (6.0%)	[33]
	Untreated/treated with different organic fertilizers (sewage sludge, biogas fermentation residue, liquid manure and dung from cattle, horses, and pigs) (Germany)	0–30	3.7 ± 11.9 [b]	Foil (61%) Fragment (28%) Fiber (1%)	1–5	PE (87%) PP (4%) Nylon (3%) PA (3%)	[35]
Irrigation water	Unirrigated/irrigated with recycled wastewater/irrigated with desalinated brackish water (Spain)	0–5	0 (non-irrigated) 159 ± 338 (irrigated with recycled wastewater) 46 ± 92 (irrigated with desalinated brackish water)	Fiber (100%)	<1	PES (25.8%) Acrylic (9.7%) PP (3.2%)	[42]
Multiple sources	Different land uses and plastic mulching (Iran)	0–10	67–400	Fiber Fragment	0.1	(-)	[25]
	Non-mulching/mulching and influence of irrigation water from nearby river (China)	0–10	262.7 (non-mulching) 571.2 (mulching)	Fragment (86.7%) Fiber Film	1–3	PE PP PES Nylon Rayon PA (fiber) Acrylic (fiber)	[44]
	Household sewage, plastic bags, nylon nets, organic fertilizer (China)	0–5	320–12 560	Microbead (48%) Fiber (37%) Fragment (15%) Foam (1%)	<0.2	PA (32.5%) PP (28.8%) PS (16.9%) PE (4.2%) PVC (1.9%)	[53]
	Different land-use, plastic mulching, greenhouse soils, irrigation water from river or groundwater (China)	(-)	3910 ± 1031 (wheat land) 5490 ± 573 (paddy land) 3683 ± 362 (woodland) 3386 ± 593 (orchard land) 5386 ± 835 (plastic mulching soil) 5124 ± 632 (greenhouse soil) Overall average: 4496 ± 1082	Fragment (54.1%) Fiber (26.9%) Film (10.2%) Sphere (8.5%)	0.02–0.2	PE (20.9%) PA (20.3%) PES (12.5%) PS (11.4%) PP (10.8%) PVC (7.8%) Acrylic (6.2%)	[54]

Table 1. *Cont.*

Potential Source	Study Area (Country)	Depth (cm)	Concentration [a]	Dominant Shape	Dominant Size (mm)	Polymers	Reference
Multiple sources	Different land-use with possible influence of compost, aeolian deposition and pipes from mining (Chile)	(-)	306 ± 360 (crop land) 184 ± 266 (pastures)	Fiber (68%)	1–2	Acrylic PU PE PP NBR PS PLA PA PES	[55]
	Conventional cultivation of vegetables whose main source of MPs is packaging bags, plastic nets, plastics/old clothes, scarecrow, rubbish bags, plastic seed trays, drums and plastic hoses for irrigation (Mauritius)	0–20	Up to 420 ± 244	(-)	<1	PP (56.2%) PA (28.7%) PE (10.2%) PS (2.7%) EVA (2.2%)	[27]
	Different land-use: cultivated land, grassland, plastic mulch covering and plastic greenhouses (China)	0–6	Up to 53.2 ± 29.7 (shallow soil) Up to 43.9 ± 22.3 (deep soil)	Film (36–41%) Fiber (21–24%) Fragment (23–27%) Foam (7–11%) Spherule (4–6%)	0.1–0.5	PE (51–59%) PA (16–23%) PS (6–8%) PP (6–9%)	[56]
	Different land-use (grassland, dry land, paddy fields and plastic greenhouses), wastewater irrigation, organic fertilizer and plastic mulching film (China)	(-)	875 ± 229–6075 ± 865 Overall average: 2522 ± 1276	Fiber Fragment	<1	(-)	[43]
	Mainly sewage sludge and irrigation water with possible inputs of garbage, river water and aeolian transport (China)	0–10	240–3660	Film (67%) Fiber (29%) Fragment (2%) Pellet (2%)	<0.5	(-)	[57]
	Different cropping characteristics (maize, sunflower, and potato farmland) and agricultural practices (plastic mulching, mechanical plowing, plastic strings, sunshade nets and clothing) (China)	(-)	11,300–78,100 [b]	Film Fragment	<0.2	PE (91.6%)	[58]
	Different land-use: facility, farmland, grassland and orchard soils with application of plastic mulching and fertilizer (China)	0–10	2795.7 (facility soil) 1860.5 (farmland) 910.9 (grassland) 1322.2 (orchard soil)	Film (18.3–91.6%) Fragment Pellet Fiber	<0.1	PE (45.5–74.3%) PP PS PVC	[59]

[a] MPs in particles kg^{-1} soil, unless otherwise noted; [b] particles kg^{-1} soil dry weight. EVA: ethylene–vinyl acetate; NBR: acrylonitrile butadiene rubber; PA: polyamide; PE: polyethylene; PES: polyester; PET: polyethylene terephthalate; PLA: polylactic acid; PP: polypropylene; PS: polystyrene; PU: polyurethane; PVC: polyvinyl chloride; (-) not mentioned or not detected.

3.2. Outcome and Behavior

3.2.1. Vertical and Horizontal Migration

In addition to being sinks, agricultural systems can also act as a dynamic MP source [15,26]. Once MPs enter agricultural soils, these particles can be transported either vertically or horizontally as a result of abiotic (wind and water erosion) or biotic (human, vegetation and soil-organism activity) factors. These transport mechanisms not only depend on the characteristics of MPs (shape, size, type, and density), but are also strongly conditioned by the properties of the soil itself. For instance, coarsely textured soils with larger pore spaces or soils with lower densities may yield higher infiltration and accumulation of MPs at deeper layers compared with finely textured or denser soils [51]. In general, it has been observed that spherically shaped particles are more easily transported through soil pores, while fibers tend to stick to soil aggregates [60]. The authors also agree that the lower the density and size of the particles (generally <1 mm) the higher their mobility [61]. Thus, high-density polymers generally migrate downward to deeper soil layers, while low-density polymers are transported horizontally over the surface by the same factors that control the mobility of natural soil particles, mainly aeolian and fluvial powers [16,61,62]. During surface runoff, the presence of vegetation can retain up to 20% more MPs because plant stalks and leaves function as a physical barrier [61], which could potentially prevent the movement of MPs into aquatic ecosystems [63]. At the root level, plants can also change the distribution depth of MPs in the soil profile. Thus, plants with predominantly primary and secondary roots tend to push MPs towards the soil surface by increasing soil porosity and providing the flotation of less dense MPs when the pores are filled with water, while plants with more tertiary roots keep MPs in the soil layers [18,49,64,65]. Infiltrated water can also cause the descent of denser and smaller MP particles into deeper soil layers via leaching [18,49,64], although only a few studies have found evidence of MPs in groundwater [42,66]. The aeolian transport of MPs in agricultural soils has also not received sufficient scientific attention, although authors warn that it could be very significant, especially in arid and semi-arid regions where particles smaller than 100 µm can be re-suspended and re-enter the environment through dry and wet deposition [25,26,67,68]. Similarly, very common agricultural practices (e.g., digging, pronging, tilling and irrigation) integrate MPs into the top and bottom layers of agricultural soils [8,15,44,64,69,70].

Soil micro- and mesofauna such as springtails, earthworms and digging mammals such as moles have been shown to play a crucial role in the transport of MPs in soils [14,71,72]. For example, earthworms transport MPs by ingesting and expelling them, by pushing them together with the litter inside the burrows they are constructing, or even because MPs tend to stick to their surface [72,73]. Smaller organisms such as collembolan species also transport MP particles in the soil environment, depending on the particle size and type and also on the size of the organism. Due to their small size, collembolans tend to have a lower distribution capacity than bigger soil fauna such as earthworms, but since they are very abundant, their impact should not be underestimated [70,71]. Beriot et al. detected MPs from plastic mulching in sheep feces, and the potential transport of MPs by a herd of 1000 sheep was estimated to be ~10^6 particles ha^{-1} $year^{-1}$ [47]. Another study with chickens feces revealed the presence of MPs due to the direct ingestion of plastic particles in soils or indirectly through earthworms ingestion [74].

Soil microbes such as fungi and bacteria can also have some impact on the migration of MPs in soils [75]. They can act as a transport vector because bacterial and fungal hyphae can be attached to the surface of extremely small MPs [15,76,77].

3.2.2. Transport Vectors for Pollutants

The impact is further exacerbated when plastics contain a variety of toxic chemicals such as bisphenols, phthalates and other short-/medium-chain chlorinated paraffins, which can comprise up to 70% of the weight of plastic [78]. However, its own toxic composition can be a threat to terrestrial ecosystems, since MP particles have shown an ability to interact with heavy metals and persistent organic pollutants (POPs) such as poly-

brominated diphenyl ethers (PBDEs), organochlorine pesticides such as dichlorodiphenyltrichloroethane (DDT) and polycyclic aromatic hydrocarbons (PAHs) such as phenanthrene (PHE), among others; these are very abundant in agricultural soils due to the broad application of pesticides [14,62,78–84]. The presence of MPs made of PE reduces the natural retention capacity of the soil, and therefore, benefits the transport of POPs, which could eventually provoke the contamination of groundwater [79]. Since MP particles show a relatively high specific surface area, they actually tend to adsorb POPs more easily than in marine environments [62]. When exposed to environmental forces (e.g., ultraviolet (UV) radiation, temperature, soil acidity and alkalinity or oxidation processes), these pollutants may be released into the soil ecosystems where they can affect soil microbial activity due to their mutagenic, endocrine and carcinogenic characteristics [62,81,85]. This aging process affects the physicochemical properties of MPs by increasing surface roughness and oxygen-containing groups, which could enhance the sorption and mobility of MPs in soils and groundwater [15,31,86]. Recent research has shown how MPs can be a vector for the transfer of heavy metals into the soil environment, causing accumulation inside soil organisms [87–89]. Soil components such as clay, silt, sand and organic matter could contribute to the accumulation of metals in MPs [31,87]. Scopetani et al. observed that soils fertilized with compost had higher concentrations of additives such as diethylhexyl phthalate (DEHP), acetyltributylcitrate (ATBC) and nonanal, which may have been transferred from plastics to compost during composting [90]. In this regard, smaller MPs (especially smaller than 100 µm) should be of greater concern in future studies, as they have a larger surface area to absorb various contaminants and increase their bioavailability [33]. In addition, more knowledge about the interactions between aging, sorption, and the transport of MPs in agroecosystems is still needed [31].

3.2.3. Degradation

As discussed above, physicochemical processes, such as photo- and thermo-oxidative degradation due to UV radiation, play a significant role in the initial breakdown and aging of the polymer structure of plastic debris in agricultural soils [89,91]. In addition, MP fragmentation can be accelerated by agricultural activities such as mechanical tillage, seeding and crop rotation [59,72], as well as by agricultural irrigation and precipitation [15,32]. The degradation of MPs is also driven by biological factors, in particular, the fungal and bacterial organisms present in soil [92,93].

4. Summary of the Principal Analytical Methods for the Determination of MPs in Agricultural Soils

As mentioned above, research on MPs in soil ecosystems is a relatively young topic, and there is not a standardized method available yet for the quantification of MPs in soils [9,28,29,40,53,81,94]. Fortunately, a variety of recent studies address this problem and focus on developing an adequate methodology to investigate and quantify MPs in agricultural soils. These analytical methods are usually accompanied by information about the type of soil and analyses of soil parameters such as soil organic matter (SOM), texture, pH, ionic strength, cation exchange capacity, aggregate stability, and bulk density; these could influence MP separation efficiency since plastic particles are strongly associated with soil aggregates [44,95].

4.1. Separation of MP Particles from the Soil Matrix

It is important to consider the three-dimensional structure of the soil during sampling. Until now, most studies have sampled the topsoil, i.e., between a 0 and 10 cm depth [19,44,94,96,97], while a lower number of them have sampled deeper layers [27,29,30,49] (Table 1). Regarding the sampling device, shovels and hoes are used for top-soil samples, whereas corers and augers are preferred for deeper soil samples, mainly of stainless steel [95]. Samples must be stored in aluminum boxes or bags, and plastic or synthetic devices and clothing should be avoided during sampling to prevent contamination [98–100].

After collection, soil samples are stored at 4 °C and air-dried [47,53,101] or oven-dried at <70 °C [29,40,56,102]. Some authors also include sieving steps (<2 mm) as a pretreatment before extracting MPs [19,29,56] (Table 2).

Extracting MP particles from soils involves a variety of physical and chemical processes that should be carefully developed. Generally, these can be categorized into sieving/filtering and density separation/flotation methods [8,35,69,103]. In many cases, an extra step is included to remove the organic material to facilitate disaggregation and MP extraction. Although this step facilitates further counting of the particles, it also bears the risk of damaging them because it usually involves a variety of chemical reactions, and therefore, could lead to erroneous results during the identification process [28,29,52].

Table 2 shows an overview of the physicochemical processes used for the MP extraction in different types of agricultural soils. A common way of extracting MPs from the soil matrix is to carry out flotation in distilled water or in high-density saturated salt solutions, such as sodium chloride (NaCl, 1.2 g cm^{-3}), sodium iodide (NaI, 1.6 g cm^{-3}), sodium bromide (NaBr, 1.4 g cm^{-3}) or zinc chloride ($ZnCl_2$, 1.7 g cm^{-3}), among others [104]. Once added to the sample, the mixture is stirred to achieve a homogenous suspension, which is then stored overnight in a clean vessel, so soil particles deposit on the bottom [19,50,94]. Impurities and floating organic and MP particles can be filtered using filter paper with a pore diameter <3 µm [19,69]. This procedure should be repeated until no visible particles are floating on the surface anymore. Zhang et al. even exposed the soil solution to ultrasonic vibrations prior to the last filtering step when they were working with polypropylene (PP) and low-density PE (LDPE) plastics [19]. After density separation, it is suggested that chemical solutions such as 30% v/v H_2O_2 or Fenton's reagent should be added to the supernatants in order to degrade all organic matter [69,94]. Afterwards the samples are filtered again and dried for further investigation [19,50,53]. Another recommended option is to remove the organic matter prior to density separation [28,42,52].

Table 2. Overview with some reviewed analytical methods about separation, identification, and quantification of MPs in agricultural soils.

Separation Method	Soil Type	Identification and Quantification Method	Ref.
Filtering and Sieving			
Wet-sieving (1 mm); transferring the residual to a Petri dishes using a squeeze bottle and spoon; removing the excess water using a disposable syringe.	Kalkmarsch; Brown earth; Pseudogley–Luvisol; Gleysol–podzol; Luvisol; Brown earth–pseudogley and Pseudogley	Stereomicroscope/FTIR	[35]
Air-drying; 5 g soil with 30 mL distilled water; stirred for 30 min at 150 rpm; centrifuging for 10 min at 3000 rpm; filtering the supernatant using filter paper (<8 mm); adding distilled water; shacking again and putting in ultrasonic bath (10 min); centrifuging again; filtering the supernatant; air-drying (24 h).	(-)	Stereomicroscope	[47]
OMR with 20 mL of H_2O_2 and deionized water (500 mL of sample); sieving (5 mm; 1 mm); repeating until all soil aggregates are dissolved (maximum 3 times).	Clay (Entisols and Vertisols)	ATR-FTIR	[96]
Dry-sieving (<2 mm); 30 g/200 mL distilled water; stirring for 1 h; wet-sieving (200–2000 µm); storing for 16 h in 500 mL distilled water; extraction of 20–50 µm; 2–20 µm and 0–2 µm fractions using Robinson's pipette method; oven-drying (60 °C); supernatant recovery: addition of saturated $SrCl_2$ solution for flocculation; centrifuging; filtering (0.45 µm).	Loam (Luvisols)	Pyr-GC-MS; visible MPs by TEM/EDXS	[29]
wet-sieving (0.5, 1, 2 and 5 mm).	Soil substrate	ATR-FTIR	[38]
Wet-sieving using a column of sieves (2, 0.25 and 0.05 mm); submerging of column in distilled water and up-/down-driving at a rate of 30 cycles per minute over a period of 5 min; drying (60 °C); centrifugation of dried samples (150 mL distilled water at 2300 rpm for 10 min); removal of supernatant; repeating the addition of distilled water and centrifugation; drying at 60 °C; OMR: adding 10 mL concentrated H_2O_2 (35%); 1 mL 10% $FeSO_4$; heating in a sand bath at 50 °C; decomposing of H_2O_2 using 10% $FeSO_4$; addition of 30 mL 0.5 M NaOH; storing for 24 h; density separation using saturated NaI solution; sieving; oven-drying at 80 °C.	Soil substrate (Nitisols; Gleysols)	Stereomicroscope	[19]

Table 2. *Cont.*

Separation Method	Soil Type	Identification and Quantification Method	Ref.
Density Separation and Flotation			
Air-drying; density separation of 100 g soil using 200 mL of saturated NaCl solution (1.19 g cm^{-3}); ultrasonic (5 min); stirred (30 min); filtering using GF/A membranes (0.45 mm); OMR with 100 mL of 30% H_2O_2 48 h at 50 °C; stirring (30 s every 2 h); vacuum filtering.	(-)	Stereoscopic microscope/μ-FTIR	[54]
Sampled using shovel; sieving: <2 mm; density separation, $ZnCl_2$ 5 M (1.55 g cm^{-3}); vacuum filtering, polycarbonate membrane.	Mollisols predominate (70%), followed by Alfisols (11%), Inceptisols (13%), Entisols (2%) and Vertisols (4%)	Stereomicroscope/μ-FTIR Microscope	[55]
Sampling using stainless-steel corer; air-dried; OMR (10 g soil with 40 mL 33% (*w*/*v*) H_2O_2 2 h at 60 °C 300 rpm; density separation using saturated NaCl solution (1.2 g cm^{-3}); filtering using stainless-steel mesh (50 μm).	Sandy-loam and clay-loam (Typic Torrifluvents and Typic Haplocambids)	Stereomicroscope/μ-FTIR	[42]
Sampled using stainless-steel corer; dried 24 h at 65 °C; OMR using Fenton's reagent: 10 mL 30% (*v*/*v*) H_2O_2 and 10 mL 5% $FeSO_4 \cdot 7H_2O$ solution at <40 °C using an ice bath; density separation: filtered RO water; saturated NaI solution (1.8 g cm^{-3}); vacuum filtering through Whatman GF-D filter paper.	Sandy-loam	Stereomicroscope for a lower size limit of 50 μm; ATR-FTIR for >300 μm (excluding fibers); μ-FTIR in transmittance mode for all smaller particles and fibers	[52]
OMR: (a) 30% (*v*/*v*) H_2O_2 at 60 °C and at 70 °C; (b) Fenton's reagent (30% (*v*/*v*) H_2O_2 with an iron catalyst consisting of 20 g $FeSO_4 \cdot 7H_2O$ in 1 L of filtered RO water and final pH of 3); (c) NaOH solution (1 M NaOH at 60 °C and 10 M NaOH at 60 °C); (d) 10% KOH solution at 60 °C; density separation: (a) use of filtered RO water to extract MPs at freshwater density (1 g cm^{-3}); (b) NaI solution (1.8 g cm^{-3}) to extract higher-density MPs; stirring for 1 min; settling for 24 h; vacuum-filtering through Whatman GF-D filter paper.	Sandy-loam (organic-rich; brown podzol soils)	Stereomicroscope/ATR-FTIR	[28]
Drying at 25 °C; density separation using saturated NaI solution (1.6 g cm^{-3}); stirring 5 min; ultrasonic for 15 min; 2 days rest to float out supernatants; filtering of supernatants; OMR with Fenton's reagent (20 mL 30% H_2O_2 and 20 mL $FeSO_4$ (0.05 M) (5 min at 60 °C); additional H_2O_2 (most 15 mL); filtering (0.45 μm GF/C glass-fiber membrane).	(-)	Stereomicroscope/μ-FTIR/SEM	[30]
Density separation using saturated NaCl solution (1.19 g cm^{-3}); drying at 70 °C for 24 h; ultrasonic treatment (2 min); stirring for 30 min to float out supernatants; settling for 24 h; filtering of supernatants using 20 μm nylon net filter; OMR with 30% H_2O_2 for 72 h at 50 °C; vacuum filtering (20 μm nylon net filters).	(-)	Stereomicroscope/μ-FTIR	[94]
Density Separation using saturated NaCl solution (1.24 ± 0.05 g mL^{-1}); stirring for 30 min; suspension for 24 h; collecting of supernatants into clean glass bottles (3 repetitions); filtering (20 μm); OMR with 100 mL of 30% H_2O_2 (72 h at 65 °C and 80 rpm); filtering (20 μm).	(-)	Stereomicroscope/μ-FTIR	[97]
Density separation using saturated NaCl solution (1.20 g cm^{-3}); ultrasonic (5 min); flotation using saturated NaI solution (1.60 g cm^{-3}) if they contain many solid particles; filtering using nylon fiber (20 μm); OMR with 30% H_2O_2 (72 h; 60 °C).	(-)	Stereomicroscope/ATR-FTIR/μ-FTIR/SEM	[34]
Sampled using stainless shovel; density separation using $NaPO_3$ (50 g air-dried soil dispersed with 0.5 mol L^{-1} $(NaPO_3)_n$ solution; Addition of saturated NaCl (flow rate of 1.0 L min^{-1}); floatation via air blowing in the bottom; collecting of the over-flow suspension; sieving of the low-density materials (50 μm); sieve residues are then settled in saturated NaI solution for 48 h; filtering of the liquid through 5 μm cellulose nitrate filter; repeating 3 times; OMR with H_2O_2 and heating (70 °C for 72 h); filtering using a 20 μm glass-fiber filter.	(-)	Stereomicroscope/μ-FTIR	[44]
Sampling using steel soil-sampler; density separation of 200 g soil using saturated NaCl solution (1.2 g cm^{-3}) for 20 min at 25 °C and 200 rpm; filtering using nylon fiber membrane (20 μm); OMR with 30% (*v*/*v*) H_2O_2 (72 h at 60 °C).	Hapli-Stagnic Anthrosols	Stereomicroscope/ATR-FTIR/μ-FTIR/SEM	[33]
Flotation using distilled water and NaI mixture; centrifuging for 10 min at 3,000 rpm; filtering (11 μm)	(-)	Stereomicroscope/μ-FTIR	[50]
Flotation using 50 mL distilled water (10 g soil) and ultrasonic treatment (2 h); filtration using filter paper (3 μm).	Clay; loess; sand	Stereomicroscope	[105]
Flotation of 200 g soil using 200 mL $ZnCl_2$ in a 500 mL glass beaker by stirring for 5–10 min and deposition for 24 h; filtering of supernatant using 0.45 μm GF/A membranes; repeating twice.	(-)	Stereomicroscope/μ-Raman spectroscope	[53]

Table 2. *Cont.*

Separation Method	Soil Type	Identification and Quantification Method	Ref.
Drying at 40 ± 2 °C; sieving (<2 mm); 5 ± 0.01 g soil with 20 mL of deionized water; stirring 30 s at ~21,000 rpm; centrifuging for 15 min at 2000 rpm; filtering the supernatant using filter paper (>8 µm); density separation using NaCl solution (1.20 g cm^{-3}); filtering; adding 20 mL ($ZnCl_2$; 1.55 g cm^{-3}); stirring for 30 s at 32,000 rpm and centrifuged for 15 min at 2000 rpm; filtering.	Loam; sand loam (Entic Haploxerolls)	Stereomicroscope	[40]
Flotation using 50 ml demineralized water (12.3 g soil); ultrasonic cleaning agitation at 50/60 Hz (2 h); settling (36 h); burning of soil samples at 120 °C.	Soil substrate (karstic)	Stereomicroscope	[74]
Flotation using distilled water; filtering (3 µm); OMR with Fenton's reagent.	Clay-loam (Mollisols)	FTIR	[69]
OMR with 30% KOH:$NaClO_4$; ultrasonic treatment with energy input at 60 J mL^{-1} for 20 min; digestion process at 50 °C for 48 h with 30 s of shaking of the samples every 2 h.; first density fractionation (3×): centrifuging at 4000 G for 5 min; addition of 30 mL saturated NaCl solution; re-centrifuging at 4000 G for 5 min; second density fractionation: $ZnCl_2$ solution.	(-)	Stereomicroscope/Raman spectroscope	[99]
Flotation using 25 mL distilled water; vacuum filtering using a fiber-free membrane.	Silt-loam; fine sandy-loam; clay-loam	Polarized-lightmicroscopy	[37]
Others			
Incubation of samples in 60 °C water bath for 6 h; suspension of sample for 24 h; extraction of a small amount of supernatant liquid which is then dried and put on a Petri dish.	Sandy-loam	TOF-SIMS	[98]

(-) No available data. ATR-FTIR: Attenuated Total Reflection–Fourier Transform Infrared Spectroscopy; EDX: Energy-Dispersive X-ray Spectroscopy; FTIR: Fourier Transform Infrared Spectroscopy; µ-FTIR: micro-Fourier Transform Infrared Spectroscopy; MP: Microplastic; OMR: Organic-matter removal; Pyr-GC-MS: Pyrolysis-Gas Chromatography–Mass Spectrometry; Ref: Reference; SEM: Scanning Electron Microscopy; TEM: Transmission Electron Microscopy; TOF-SIMS: Time-of-Flight Secondary Ion Mass Spectrometry.

Based on the literature consulted in this review, NaCl solution and distilled water are the most-used in density separation (26.1% and 17.4%, respectively), although 34.8% of the authors apply various solutions [40,44,49,50,106]. Around 26.1% of the authors do not include organic-matter treatment steps, compared to 52.2% who prefer the use of aqueous H_2O_2 and, to a lesser extent, Fenton's reagent (13.0%). In this regard, Radford et al. showed that both reagents remove similar amounts (>70%) in soils with low organic content, although aqueous H_2O_2 obtained better results in soils with high organic content [95,104].

Another method is sieving the samples, either dry or wet, in a column of sieves with various mesh sizes [19,29]. After dry-sieving the soil sample, the matter is mixed with distilled water and sieved again, wet. Large particles will float on the surface of the solution and can be removed easily. Sieving should be repeated using various grid sizes to obtain different fraction sizes. Fractions then can be oven-dried and weighed afterward. In order to receive the most reliable results, the supernatants should be filtered and organic matter can be removed if needed [29,40]. Recently, other emerging methodologies have also been developed for the extraction of MPs using different oils for density separation and enzymatic and oxidative digestion for organic-matter removal [95].

4.2. Identifying and Quantifying MP Particles

After separation, samples can be analyzed via visual sorting, i.e., stereomicroscopy, and spectroscopic techniques, such as micro-Fourier transform infrared spectroscopy (µ-FTIR), Raman microscopy, scanning electron microscopy (SEM) or thermogravimetry−mass spectrometry (TG-MS), to identify and quantify MPs (Table 2) [19,29,40,53,82,89,94,107,108]. To make it easier to distinguish between natural particles (e.g., aluminum silicate or quartz) and MP particles, the samples are sometimes heated between the observations under a microscope (hot-needle test), which is usually connected to a camera [19,50]. On the basis of spectral analyses, the polymer types can be detected within the samples [107]. There are several software packages such as ImageJ, OMNIC™ and OPUS that work with polymer databanks to detect MPs in images [40,94,96]. Then, particles are classified according to their polymer type or morphological characteristics (shape, color, and size); however, since

there is no standardized protocol for the analysis of MP particles, the classification of MPs is very heterogeneous (see Table S2 of the Supplementary Material as an example).

The great majority of the reviewed studies determined the number of MPs in agricultural soil samples via counting, and expressed them as number of particles kg^{-1} soil [19,38,50,53,69,74,94,96,97,99]. In some cases, the counts were used to estimate weights by taking into account the density of the polymers [19,40,48,69].

Even though the number of particles per kg of soil is the dominating quantification unit for MPs in agricultural soils, the variations in methodology make comparisons complicated. On the one hand, some studies used dry mass, whereas others used wet mass (see Table 1). On the other hand, quantifying MPs in terrestrial environments can be complicated since visual identification always bears the risk mistakes being made during counting [40]. Furthermore, some of the suggested separation methods are likely to not only destroy the organic material of the soil matrix but also break down MPs [28,29].

4.3. Contamination Control

Strict quality assurance and quality control (QA/QC) procedures are required to avoid any possible contamination during field sampling and MP analysis in the laboratory [44]. Although there is no standard protocol for the prevention of contamination, a series of common recommendations have been established. All material must be plastic-free (e.g., glass or metal materials) and preferably covered to avoid airborne contamination. The equipment and materials in the laboratory should be cleaned using Milli-Q®, distilled, deionized or ultrapure water before use and rinsed before and after contact with each sample to prevent cross-contamination [109]. Cotton clothing and nitrile gloves should always be worn during analysis [95]. Other authors go even further and carry out the analytical processes under a laminar-flow hood and pre-filter all the employed reagents [52,53,56]. In addition, it is necessary to realize blank samples performed in parallel to evaluate systematic errors in the experiment [109].

5. Occurrence of MPs in Agricultural Soils

As can be seen in Table 1, the abundance of MPs is closely related to soil management. Thus, areas with elevated exposure (multiple sources identified) showed the highest concentrations of MPs [27,44,53–55]. However, these vary substantially depending on the location. For instance, Chen et al. reported a concentration between 320–12,560 particles kg^{-1} in vegetable fields in China, while Ragoobur et al. found a mean of 320 ± 112 particles kg^{-1} in conventional vegetable fields in Mauritius at a depth of 0–10 cm in both cases [27,53]. Nevertheless, the meta-analysis data only serve as reference since the methods of MP extraction and quantification differed considerably between studies [33,34,110]. In addition, the discrepancy in contamination levels may be related to differences in cropping characteristics (e.g., cereal or root crops) and agricultural practices (e.g., plastic mulching, mechanical plowing and residue retention), even within agricultural soils in the same region [43].

All soils with plastic mulching application contained higher amounts of MPs than soils without mulching [40,47,51]. This is also supported by Ren et al., who reported that mulching films contributed 10–30% to MP contamination in soils from 19 provinces in China [31]. Studies with long-term application periods concurred that the abundance of MPs increased over time [30,47,49].

A predominance of microfibers (up to 97% or 100%) is mainly associated with soils with a history of sewage-sludge application [33,40,50,51] or soils irrigated with wastewater and water from contaminated rivers [42,44] (Figure 1). The reported size fractions varied widely across studies (see Tables 1 and 2). The great majority of the studies observed small particle sizes (generally <1 mm) in agricultural soils, with minimum detected MP sizes of >25 µm [50,52,98,111]. Regarding the types of microplastics, the polymers mostly detected were PE and PP, which were present in 41.4% and 37.0% of the studies analyzed, respectively, followed by polyvinyl chloride (PVC, 19.6%), polystyrene (PS, 17.4%),

polyamide (PA, 15.2%), polyester (PES, 13.0%), acrylic (8.7%), polyethylene terephthalate (PET) and nylon (4.3% in both polymers) (Figure 2).

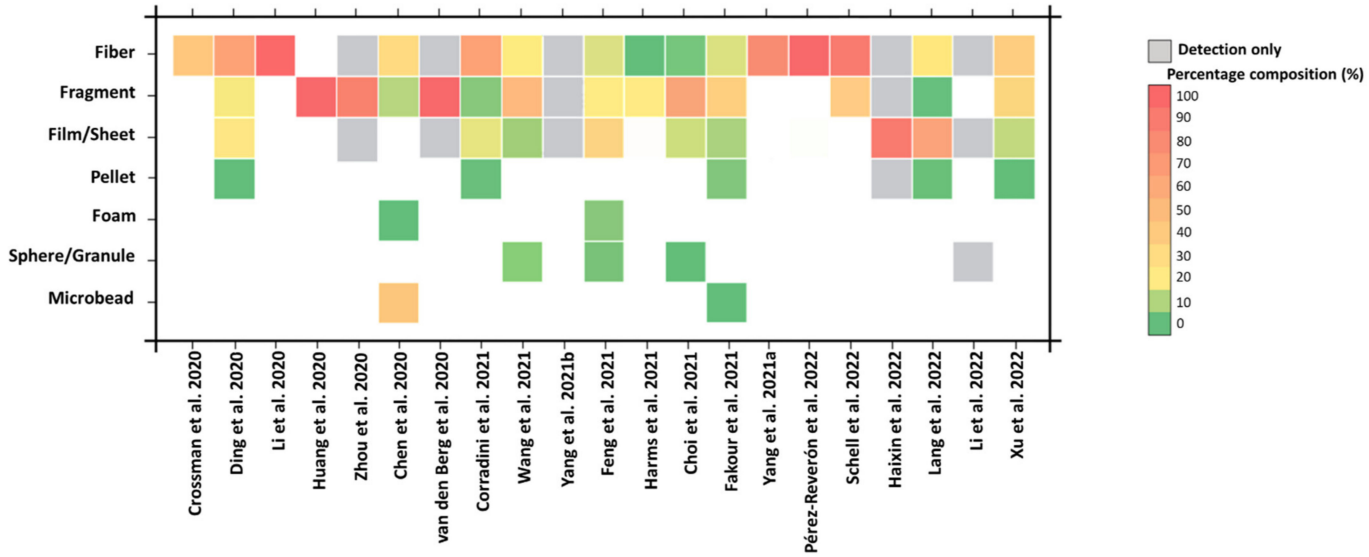

Figure 1. Percentage of different shapes of MP particles from agricultural soils.

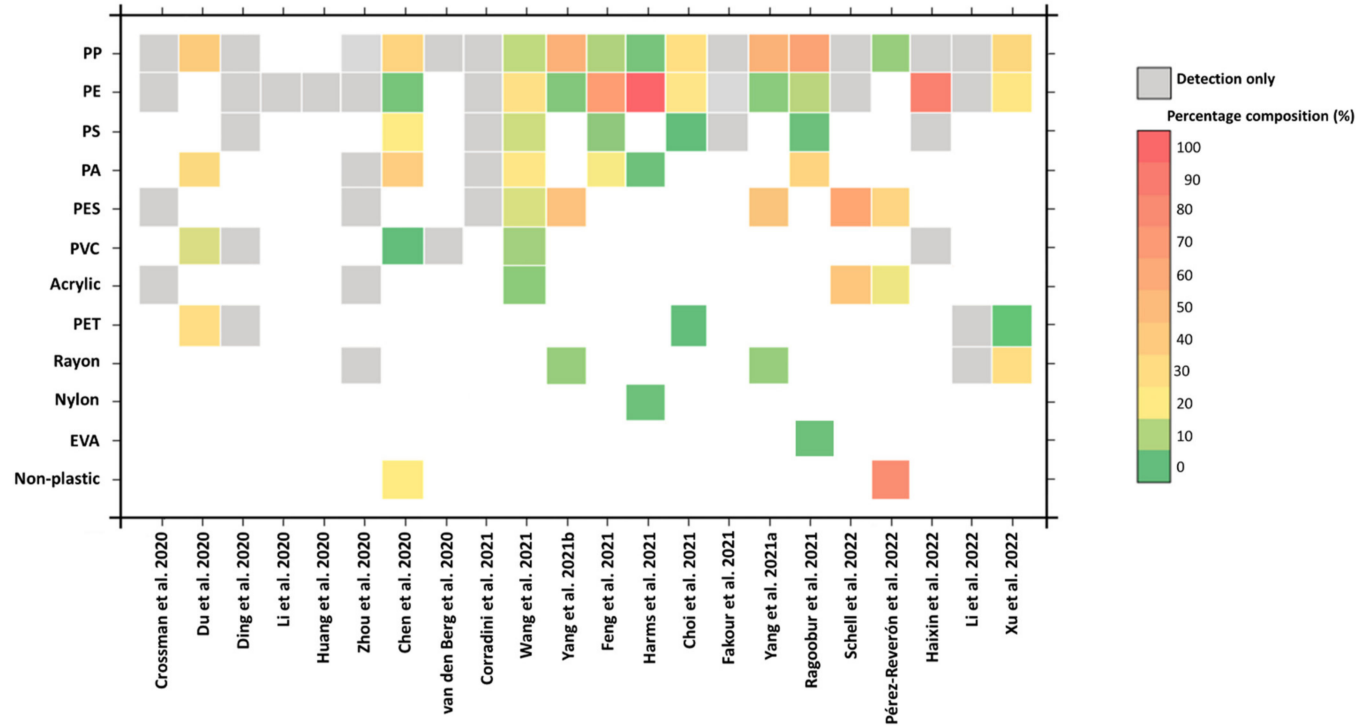

Figure 2. Percentage of different polymers of MP particles from agricultural soils. EPS: Expanded Polystyrene; EVA: Ethylene–vinyl acetate; PA: Polyamide; PE: Polyethylene; PES: Polyester; PET: Polyethylene terephthalate; PP: Polypropylene; PS: Polystyrene; PVC: Polyvinyl chloride.

The few studies on vertical distribution suggest that MPs tend to accumulate in shallow soil [49,112]. Conversely, Crossman et al. observed an increase in MPs in deeper soil layers [52]. Schell et al. explain that these discrepancies probably stem from soil properties and could influence the infiltration process, especially soil texture and structure [51]. In this regard, the investigations of Yu et al. showed that MP abundance correlated negatively with the silt fraction and positively with the sand fraction [113]. Similarly, the concentrations of

MPs varied between two types of soil (sandy-loam and clay-loam) with the same treatment (238.3–91.1 particles kg^{-1} vs. 79.5–0.0) [42].

6. Effects of MPs in Agricultural Soils

6.1. Soil Physicochemical Properties

The accumulation of MP particles can affect important soil physical and chemical properties and processes, such as bulk density, porosity, specific surface area, pH, cation exchange capacity (CEC), nitrification, hydraulic characteristics and evaporation [12,22,77,114,115]. Depending on their shape, type, size and quantity, MPs can reduce or increase water-stable aggregates, which leads to changes in soil structures that are important for soil ecosystem functioning [20,22,53,115–118]. For instance, after the exposure of PE to soils (0.1% *w*/*w*), a decrease in both the pH and the aggregate mean weight diameter (MWD) was observed. A free exchange of protons in the soil water is suggested since MPs' high surface area may affect soil CEC, and a reduction in large macro-aggregates may indicate alterations in the aggregate bindings mechanisms, directly affecting the soil structure stability and its resistance to erosion processes [115]. Moreover, this disruption of soil stability due to the presence of MPs may also affect the soil's ability to transport nutrients and its chemical properties. As an example, the addition of PP to soils (28% *w*/*w*) increased the nutrient contents of dissolved organic matter (DOM), including humic and fulvic acids. An increase in these aromatic organic compounds with high molecular weights may facilitate the transport and bioavailability of soil and/or MP pollutants. However, improvement in the DOM quality may also stimulate soil enzymatic activity, and therefore the nutrients availability for plants [114]. By contrast, several authors have also suggested that MPs can decrease soil fertility and alter the functions of the microbial community and the C, N and P cycles, especially the turnover and decomposition of organic matter in soil ecosystems, leading to important potential ecological risks [117,119]. Thus, it is generally concluded that further research is needed to delve into the mechanistic pathways of how MPs affect soil properties and, consequently, how they interact with biotic soil components (vide infra) [22,114,115].

6.2. Soil Microbiota

Due to their ubiquity and their relatively big surface area, MP particles represent a new ecological niche for bacterial communities in soil environments, which is known as the terrestrial plastisphere [120]. The composition of the accumulating bacteria and their effect on the soil biota depend not only on the characteristics and concentration of MPs, but also on the environmental conditions of the soil matrix [16,20,22]. Changes in soil porosity might alter oxygen flows and, therefore, influence the distribution of aerobic and anaerobic microorganisms [77]. Furthermore, recent studies indicate that the abundance of MPs in soils has an increasing effect on the nutrient contents of dissolved organic matter; therefore, they stimulate microbial activity in soils [9,20,114,116]. Some of these evolving bacterial communities might actually have the potential to degrade MPs and reduce levels of toxic agents and heavy metals [20], and some soil fungi have also been shown to contribute to the biodegradation of MPs [114]. Zhu et al. proved that additives as phthalate esters alter the soil microbial system and nitrogen cycling by increasing soil NH_4^+ and decreasing NO_3^- content [121]. However, in others studies, the presence of MPs decreased microbial richness and diversity or did not have a very significant impact [122]. These contradictory findings underline the need to conduct further research under different scenarios.

6.3. Soil Fauna

Due to their small size, MP particles are likely to be ingested by soil meso- and microfauna. They represent one of the lowest levels of the trophic food chain, making them a potential entry for MPs into ground-dwelling animals and even humans [6,9,53,77]. Soil fauna exposure to MPs depends on various factors such as the type and size of the species, their feeding strategy, ecological niche and time of exposure, and the quantity, size, shape and polymer composition of the MPs [123–126]. MPs can harm soil fauna in

a physical way (i.e., external skin damage and internal injuries of the digestive organs caused by direct ingestion) and also in a biochemical way (e.g., disruption of lipid, osmotic and carbohydrate metabolisms) [127,128]. Many of these harmful effects are not directly linked to the MPs themselves, but to poisonous substances adsorbed such as POPs, which provoke infertility, alterations in growth, and hepatic and oxidative stress in soil fauna [6,9,44,53,77,82,129–133]. Moreover, soil organisms play a crucial engineering role in edaphic ecosystems, so MPs could also alter bio-geophysical processes and plant growth [115,134,135]. Due to the relatively high number of individuals, their sensitivity towards environmental changes and their ecological value, many soil-inhabiting species serve as bioindicators for observing anthropogenic contamination [123,136].

Soil-inhabiting worm species such as *Lumbricus terrestris*, *Eisenia fetida* and *Enchytraeus crypticus* represent a well-studied group regarding the interactions between MPs and soil fauna. When exposed to MPs, earthworms do not necessarily show higher mortality rates, although the ingestion and accumulation of MPs potentially contaminated with pollutants could lead to a decrease in survival rates in highly contaminated soils [6,44,53,74,115,124,128,137]. According to Lahive et al., PVC has a negligible effect on reproduction in *E. crypticus*, while nylon reduces their reproduction rates by 25% [125]. Ma et al. showed that PVC and PA, in combination with other common pollutants in soils such as antibiotics, reduce reproductivity and alter microbial communities within *E. crypticus* [136]. Despite the fact that MP contamination generally represents a threat to earthworm species, the amount of biodegradable plastics could be reduced during vermicomposting [19,138], although the interaction between this type of biodegradation and POP-enriched MPs is still unknown [139]. Sforzini et al. also suggested that, in contrast to conventional MPs, exposure to biodegradable plastics did not affect earthworms' health [136]. However, these species also have an impact on the distribution of MPs in the soil profile, which could contribute to MPs' bioavailability [19,130,140].

Nematodes are also excellent study species because the detection of MPs in their transparent bodies is relatively easy [123,141]. Nematodes are likely to take up MPs in benthic sediments and soils because they confuse the particles with food [71,142]. In nematodes, the ingestion of MPs leads to decreases in intestinal calcium levels and causes oxidative stress, provoking alterations in energy metabolism that are potentially associated with higher mortality and lower reproduction rates [71,141,143]. Bacterial-feeding nematode species with relatively fast reproduction rates and high nutrient demands seem to be more susceptible than species with a slower life cycle [123,125,126].

Land snails *Achatina fulica* could probably also serve as bioindicators for MP contamination in soils because they are able to take up MP fibers, which do not lead to mortality but harm their digestive systems and influence their feeding and excretion behavior [144]. It was also shown that during ingestion and digestion, the morphology of the fibers was altered, confirming similar observations made by analyzing earthworms [144]. This supports the assumption that soil fauna potentially contributes to the chemical and physical degradation of MPs.

Another comparable well-studied group is formed of soil microarthropods such as springtails [71,131,145]. According to Barreto et al., the species richness and community composition of Collembola, Acari (Mesostigmata, Prostigmata, Astigmata) and other invertebrates are not affected by MP abundance [145]. It was found that soil-dwelling microorganisms such as springtails show lower mobility, changes in feeding behavior and a reduction in growth and reproduction [71,131,146]. Springtails seem to prefer MP-free environments over contaminated soils, which means that high MP contamination potentially modifies their movement patterns [146]. Furthermore, the effects of MP contamination seem to provoke changes in the gut microbial community of *Folsomia candida* [146]. Rondoni et al. indicated that elevated concentrations of MPs in soils can indirectly interfere with the behavioral response of fungus gnats seeking an oviposition female substrate, affecting plant–herbivore trophic interactions, with implications for crop production [147].

Soil-dwelling vertebrates represent a barely studied faunistic group compared to invertebrates. Deng et al. proved, in a laboratory experiment with mice (*Mus musculus*), that MP exposure had similar effects on their metabolic activity and digestive system to those found for earthworms, especially when exposed to high doses of MPs [148]. Therefore, the accumulation of MPs in mice seems to alter neurotic responses, energy and lipid metabolisms, and provokes oxidative stress [127,148].

Finally, the evident impact of MPs on soil fauna led Ferreira-Filipe et al. to recommend the evaluation of MPs effects on different organisms in ecotoxicity tests for the certification of plastic products, as the processes applied nowadays might be overly permissive and ignore essential factors for the health of soil species [149]. The effect of MPs on soil fauna depends largely on their concentration, which stresses the importance of the quantification of MPs in soils [6,125]. Natural environments remain a relatively unstudied field and, until now, many quantification methods have not been able to detect MPs < 100 µm; this means that the current observed concentrations might be underestimated, hence the impact on soil fauna [125].

6.4. Vegetation

There are not yet many studies about the impact of MPs in soils on vegetation. Among those that have been carried out, Boots et al. found that the seeds of perennial ryegrass (*Lolium perenne*) seem to germinate significantly less when exposed to polymers. Moreover, the chlorophyll-a/chlorophyll-b ratio of *L. perenne* increased when seeds were exposed to MPs; this may be because the photosynthetic capacity was affected by the alteration in nutrient availability induced by the presence of MPs [115]. In this regard, polymers such as PAs, nylons and acrylics contain nitrogen atoms, which could yield an increase in foliar nitrogen amounts [150], and therefore, different responses of nitrogen-dependent chlorophyll content [115]. Gentili et al. studied the toxic impact of PVC on the weed species *Senecio inaequidens* and *Centaurea cyanus* for about two months, finding significant changes, compared to control plants, in growth (lower width for both weeds and lower height for *C. cyanus*), phenology (around two days of delay for the second leaf emergence in *S. inaequidens*) and photosynthetic efficiency (lower for *C. cyanus*) [151].

Other experiments on *Plantago laneolata*, *Leucanthemum ircutianum*, *Prunella vulgaris* and *Festuca guestfalicia* also demonstrated that polymers have a negative effect on plant growth [152]. Kleunen and coworkers observed that *P. laneolata* exposed to higher concentrations of an ethylene–propylene–diene monomer (EPDM) showed significant decreases in biomass, root length and survival chances. Surprisingly, they also found that low quantities of EPDM granules slightly increased biomass [152]. The actual reason that MPs affect plant growth is not very clear yet, but it is assumed that the plants' response to MPs might be related to their hydrophobic characteristics and the effect of some additives in the polymers [115,152]. Drought is another abiotic factor that may interact with MPs to induce changes in plant behavior, as highlighted by Lozano et al. [153]. They found that a plant community formed of seven grass and herb species suffered shoot- and root-mass decreases under drought conditions, although they increased when PES microfibers were present. However, at the ecological level, it should be remarked that some species became more dominant due to the MPs' presence [153].

It is important to highlight those micro-sized plastics are usually too big to be taken up by plants. However, if they break down to nano-sized plastics, they might be taken up by some plant species, even at a very early growth stage (<7 days after sowing); this could impact their self-development and pose a major hazard due to being distributed up the food-chain system through edible plants [9,50,154,155].

6.5. Agricultural Production

Soil MPs may cause direct damage at early crop stages via physical blockage of the pores in the seed capsule or in roots [12]. Indeed, very short-term negative effects on edible plants' growth may appear just 6 days from seeding under MP exposure (Table 3) [21].

Furthermore, the high stability of polymeric plastic molecules highly complicates their degradation processes; even compostable plastic bags remain unaltered after being buried in soil for 27 months [156]. Although the persistence of MPs in soils and their long-term effects are difficult to assess [157], it is well known that the growth of agriculturally interesting plants may be affected by the direct uptake of MPs from soils via apoplastic (uptake via passive diffusion) and symplastic (uptake via osmosis) pathways, and subsequent transport to the whole plant via the vascular system [83]. Moreover, the tiniest plastic particles may even enter the plants through stomatal foliar uptake (Figure 3) [158]. As discussed above, MPs also indirectly affect plant behavior due to the modification of soil properties, nutrients and microbiota [83]. In any case, from an agricultural production perspective, MPs show, in general, a negative impact on crop productivity [159–161], although beneficial effects on plants growth have also been reported [160].

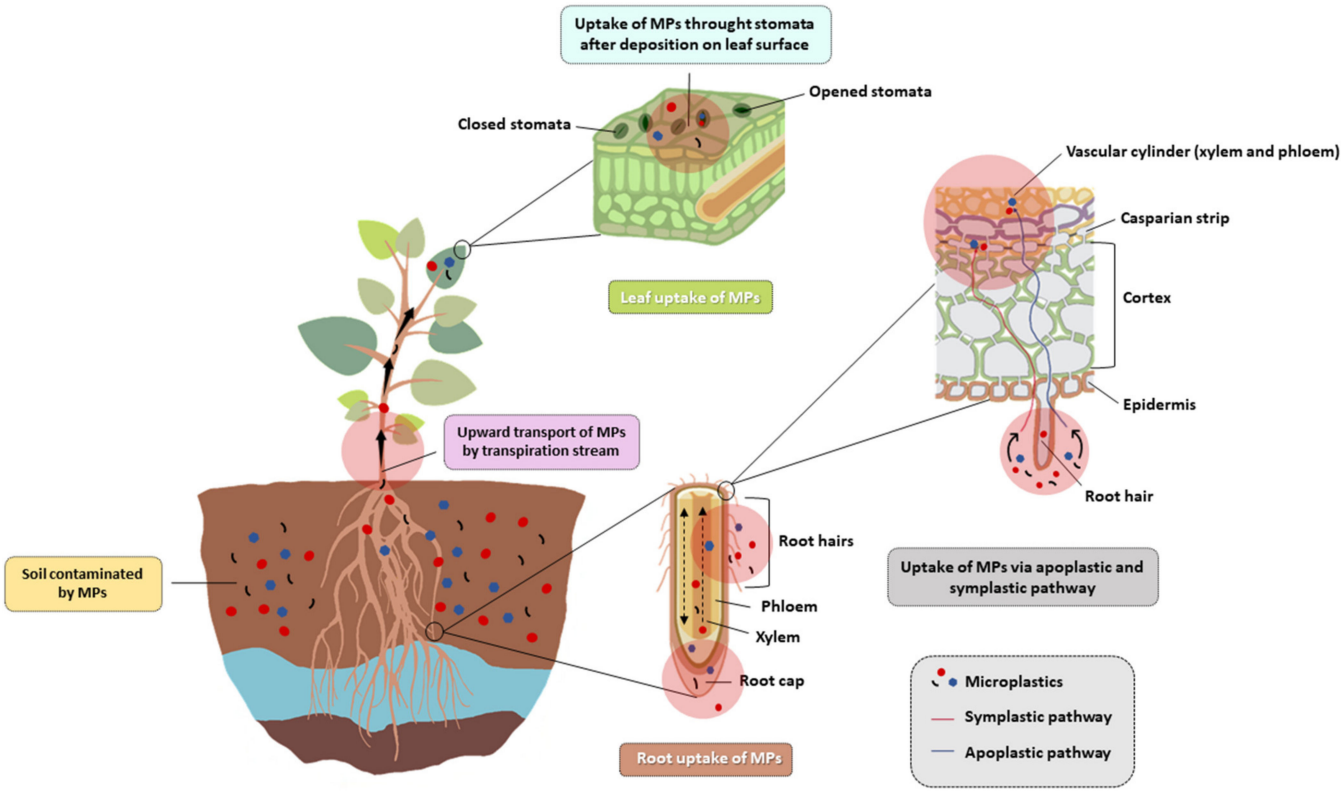

Figure 3. Uptake and translocation mechanisms of MPs in plants.

Table 3 summarizes the significant effects observed in several growth parameters of diverse crops exposed to soils polluted with MPs (for a selection of studies about the impact of MPs in crops grown with no soil, see Table S4). In general, commonly measured agronomic parameters such as plant height, fresh and dry aerial and roots biomass, root length and diameter, and seed germination rate show a tendency to decrease under MP exposure; however, sometimes, an increase was found (see Table 3). As discussed below, these data should be carefully handled because within the same crop, a given parameter may show noteworthy differences depending on factors such as the cultivar; measurement date; composition, size, dose, and aging period of the MP; location and experimental design; and presence of other co-factors (see Table 3).

For instance, when Lian et al. measured plant height and aerial dry biomass in 6-week-old maize (*Zea mays*) exposed to 1% polyurethane (PU), they found an increase in cv. ZTN 182, whereas cv. ZNT 488 remained identical to the control [162].

The measurement date is also a factor that must be kept in mind when comparisons between different reports are tackled. For instance, the exposure of PP or PVC to cress

(*Lepidium sativum* L.) led to an increase in fresh aerial biomass at 6 days, although this parameter decreased when measured at 21 days [21].

The nature of the MPs is a crucial factor, as reported Meng et al., who studied common bean (*Phaseoulus vulgaris* L.) behavior in the presence of low-density PE and a biodegradable plastic consisting of poly-butylene-adipate-co-terephthalate (PBAT)/polylactic acid (PLA)/calcium carbonate in a 85/10/5 ratio [163]. They found that at 105 days after seeding, the full mature-fruit biomass decreased when plants were exposed to 2 and 5% PBAT + PLA bioplastic, but not under PE exposure. A similar trend was observed in other parameters such as shoot and root dry biomass, measured at the end of the vegetative stage (46 days) [163].

MPs size may be also a determining factor, e.g., cress (*L. sativum*) shoot height decreased when seedlings were treated with 6–60 and 500–3000 µm PET, but not with the intermediate size of 61–499 µm [164]. On the contrary, the three size ranges led to the same response in other measured parameters: the leaf number decreased, the inhibition-of-germination percentage increased and total seedling fresh biomass remained equivalent to the control [164].

MPs dose importance can be deduced from the research of Wang et al. with lettuce (*Lactuca sativa* L.) [165]. They reported that aerial and radicular dry biomass decreased in three different soils containing 10% PE, but no significant changes were found in comparison to the control when PE doses were set at 0.1% and 1% [165].

The period that MPs particles retain their nature while subjected to aging processes also plays a role in crop production. Pflugmacher et al. mixed artificially aged (0, 40, 80, 120 and 160 days through a hygrothermal accelerated method) polycarbonate (PC) with soils to grow cress (*L. sativum*) [166]. It was discovered that the negative effect of PC on fresh and dry root, shoot and seedling weights decreased with time, and no differences were found between the PC aged for 160 days and the control [166].

Most of the discussed experiments were developed in highly controlled conditions such as laboratory climate-controlled chambers [84,162,164,166–169] or greenhouses [150,163,164,170–172], but field experiments to evaluate the effects of MPs in extensive farming are still scarce [173–175]. Data collected from small-scale trials might differ from those obtained from large-scale experiments. For instance, on the one hand, Li et al. found that barley (*Hordeum vulgare* L.) grown for 14 days in hydroponic boxes and treated with PS showed a decrease in rootlet number as well as an increase in oxidative stress and metabolic-damage indicators (see Table S4 for further details) [176]. On the other hand, Greenfield and co-authors evaluated, via an eight-month field experiment, the effects of the expected amount of MPs generated during 1–3 years of plastic-mulch-film application in a winter barley crop [174]. The application of PE and biodegradable poly(3-hydroxybutyrate-co-3-hydroxyvalerate (PHBV) yielded no differences in the agronomic parameters of interest (e.g., grains per ear and straw, and grain and ear weight) in bacteria or archaeal diversity, although N_2O emissions and soil moisture decreased when conventional plastic was used [174]. Hernández-Arenas et al. also performed an outdoor experiment consisting of growing tomato plants (*Lycopersicon esculentum Mill.*) in pots using soils fertilized with sludge containing MPs, finding that MPs increased growth but delayed and decreased fruit production [175].

Some co-factors have been evaluated together with MPs. It has been found that MPs can aggravate the harmful effect of other co-pollutants in crops (heavy metals [83,165], inorganic nanoparticles such as ZnO [168], organic compounds such as PHE [84,171], etc.) and of other pernicious phenomena such as acid rain (AR) [164]. By contrast, according to Qi et al., the presence of earthworms may help to mitigate the toxicity of MP particles both from low-density PE and from a starch-based bioplastic in wheat (*Triticum aestivum* L.) crops [167]. For instance, they found that the anecic earthworm *Lumbricus terrestris* prevented a decrease in root dry biomass after vegetative growth (two months) [167].

In addition to the above-discussed influence of MPs on growth parameters, their correlation with mineral-element content has also been studied, although to a lesser extent [150,169]. PA increased the leaf N content in spring onion (*Allium fistulosum* L.),

whereas it was decreased by PES [150]. Colzi et al. performed an extensive analysis of element contents (K, Ca, Mg, Fe, Zn, Cu, Mn and Ni) in the roots, stems and leaves of zucchini (*Cucurbita pepo* L. var. Faenza) independently exposed to three doses of PE, PVC, PP and PET, finding differences with their respective control samples, except in stem Mn content [169].

Table S3 of the Supplementary Material expands the data of Table 3 and reflects the biochemical effects of MP exposure in several crops. Briefly, the presence of MPs generally yields higher values of oxidative stress indicators such as superoxide anion (O_2^-), H_2O_2 and malondialdehyde (MDA) [21,164,170]. Consequently, antioxidant enzymes (AOE) such as superoxide dismutase (SOD), catalase (CAT) and peroxidase (POD) normally exhibit higher levels [84,170,171]. In general, exposure to MPs leads to lower chlorophyll and carotenoid contents [21,163,166,169,172], although in some cases, no significant changes are observed [167,169,171,173], and increases can even be found [84]. These contradictory findings side with those previously discussed about crop growth; therefore, they emphasize the need for further research, especially considering the serious damage that MPs may cause to an agriculture-based economy, as well as the human health risks due to their transfer to the food chain [177].

Table 3. Significant increases (↑) and decreases (↓) observed in selected growth parameters of plants of agricultural interest exposed to MPs compared to control plants.

Species	MP	Size (µm)	Dose [a]	Measurement Date (Days)	Number or Leaves	Leaf Area	Height	Fresh Aerial Biomass	Dry Aerial Biomass	Bulb Dry Biomass	Bulb Water Content	Fresh Root Biomass	Dry Root Biomass	Dry Root/Leaf Biomass	Root Length	Root Diameter	Root Tissue Density	Seed Germination Rate	Co-Factor	Reference
Barley	PE	40–48	0.01	240			=		=											[174]
Barley	PHBV	1–15	0.01	240			=		=											[174]
Bean	PE	250–1000	0.5, 1, 1.5, 2, 2.5	46		[c]			=				=	=						[163]
Bean	PLA + PBAT	250–1000	0.5, 1, 1.5, 2, 2.5	46		↓[c]			↓[c]				[c]	↑[c]						[163]
Chinese cabbage	PE	<25, 25–48, 48–150, 150–850	0.25, 0.5, 1, 2	30				=												[172]
Chinese cabbage	PS	<25, 25–48, 48–150, 150–850	0.25, 0.5, 1, 2	30				↓[c,d]												[172]
Cress	Aged-PC	3000	2	7			↓[e]	↓[e]	↓[e]			↓[e]	↓[e]		↓[e]			↓		[166]
Cress	PE	<125	0.02	6, 21			↓,↓	=,↓										↓,↓		[21]
Cress	PE + PVC	<125	0.02	6, 21			↓,↓	↑,=										↓,=		[21]
Cress	PET	5–60, 61–499, 500–3000	0.02	6	↓		↓[d]											↓	AR	[164]
Cress	PP	<125	0.02	6, 21			↓,↓	↑,↓										↓,↓		[21]
Cress	PVC	<125	0.02	6, 21			↓,↓	↑,↓										↓,=		[21]
Lettuce	PE	8.68–500	0.1, 1, 10	45					↓[c]				↓[c]						Cd	[165]
Lettuce	PVC	0.1–18, 18–150	0.5, 1, 2	21											↑[c,d]	↑[c,d]				[170]
Maize	PE	100–154	0.1, 1, 10	30					↑[c]				↑[c]						ZnO	[168]
Maize	PLA	100–154	0.1, 1, 10	30					↓[c]				↓[c]						ZnO	[168]
Maize cv. ZNT 488	PU	4280	0.01, 0.1, 1	54			=		=				=							[162]
Maize cv. ZTN 182	PU	4280	0.01, 0.1, 1	54			↑[c]		↑[c]				=							[162]
Rice	PS	8.5–30.7	0.005, 0.025	142			↓	↓				=			=					[173]
Spring onion	PA	15–20	2	40				↑		↓	↑			↓	↑	↓	↓			[150]
Spring onion	PE	643	2	40				=		↑	=		↑	↑	↑	↓	↑			[150]
Spring onion	PES	5000	0.2	40				=		↑	↓		↑	↑	↑	↓	↑			[150]
Spring onion	PET	222–258	2	40				=		↑	↓		↑	↑	↑	↓	=			[150]
Spring onion	PP	647–754	2	40				=		↑	↓		↑	↑	↑	↓	=			[150]
Spring onion	PS	547–555	2	40				=		↑	=		↑	↑	↑	↓	=			[150]

Table 3. *Cont.*

Species	MP	Size (µm)	Dose [a]	Measurement Date (Days)	Number or Leaves	Leaf Area	Height	Fresh Aerial Biomass	Dry Aerial Biomass	Bulb Dry Biomass	Bulb Water Content	Fresh Root Biomass	Dry Root Biomass	Dry Roo/Leaf Biomass	Root Length	Root Diameter	Root Tissue Density	Seed Germination Rate	Co-Factor	Reference
Tomato	PET	310–2110	17,870, 27,821, 47,130 [b]	109			=		↑[c]				↑[c]		=					[175]
Wheat	PE	200–250	0.5, 1, 2, 5, 8	15			=	↑[c]				↑[c]			↑[c]				PHE	[84]
Wheat	PE	50–1000	1	61	=	=	=		=				↓						EW	[167]
Wheat	Starch	50–1000	1	61	↓	↓	=		↓				↓						EW	[167]
Zucchini	PE	40–50	0.02, 0.1, 0.2	28		↓[c]		=	↓[c]			=	=							[169]
Zucchini	PET	40–50	0.02, 0.1, 0.2	28		=		↓[c]	↓[c]			↓[c]	↓[c]							[169]
Zucchini	PP	40–50	0.02, 0.1, 0.2	28		=		↓[c]	↓			↓[c]	↓[c]							[169]
Zucchini	PVC	40–50	0.02, 0.1, 0.2	28		↓		↓[c]	↓			↓[c]	↓[c]							[169]

[a] % *w*/*w* MP/dry soil weight unless otherwise noted; [b] particles kg^{-1} dry weight; [c] MPs dose-dependent; [d] MPs size-dependent; [e] MPs aging-period-dependent; AR: acid rain; cv: cultivar; EW: earthworm; MP: microplastic; PA: polyamide; PBAT: poly-butylene-adipate-co-terephthalate; PC: polycarbonate; PE: polyethylene; PES: polyester; PET: polyethylene terephthalate; PHBV: poly(3-hydroxybutyrate-co-3-hydroxyvalerate); PHE: phenanthrene; PLA: polylactic acid; PP: polypropylene; PS: polystyrene; PU: polyurethane; PVC: polyvinyl chloride.

7. Conclusions and Research Gaps

MP contamination has manyeffects (mainly negative), not only on well-studied aquatic environments, but also on terrestrial ones. The research history of the impact of MPs on agricultural soils is relatively short, and the number of conducted studies is limited. Almost all of the reviewed studies are very recent and were published after 2015, with some exceptions. It can also be asserted that the study areas are restricted to very few countries. Most of the experimental studies were executed in China, which is to be expected because it is the main plastic producer worldwide. Many of the research works addressed various topics at the same time. The investigations on the possible effects of MP contamination in agricultural soils can further be distinguished into impact on transportation, soil fauna, vegetation growth, microorganisms and bio-geophysical and chemical processes.

There is still no standardized methodology regarding the quantification of MPs in soil environments, so comparisons—for example, between different agricultural soil types, agrosystems or ecoregions—are, therefore, difficult.

This review reveals that MP particles could manipulate crucial bio-geophysical processes, harm soil micro- and mesofauna, disrupt microbial activity and affect plant growth. However, considering the limitations of the reported experiments, the prospects of analyzing the interactive effects of MP size, polymers, aging and migration processes should remain in further research. For example, it is still necessary to study how MPs influence the interactions between the members of different soil trophic levels. Although most of the results point to negative effects of the presence of MPs on crop productivity, the very limited number of studies in this regard and, above all, the lack of studies under field conditions, make it impossible to establish the threshold contamination rate that could generate a significant decline in agricultural yield. The evaluation of the impact on crop production through field experiments or the use of mesocosms could be used as a bioindicator that encompasses the multiple potential effects of microplastics in the agricultural soil system.

Supplementary Materials: The following supporting information can be downloaded at: https://www.mdpi.com/article/10.3390/agriculture12081162/s1, Table S1: Analyzed effects of MPs in the consulted research articles; Table S2: Overview of some reviewed classifications of MP particles in agricultural soils; Table S3: Significant growth and biochemical effects observed in MP-exposed plants of agricultural interest compared to control plants; Table S4: Significant growth and biochemical effects of selected experiments in which MPs were exposed to hydroponic or Petri dish-grown plants of agricultural interest compared to control plants.

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