Environ. Eng. Res. 2023; 28(6): 220716
https://doi.org/10.4491/eer.2022.716

pISSN 1226-1025
eISSN 2005-968X

Review

Effect of microplastics on soil microbial community and microbial degradation of microplastics in soil: A review

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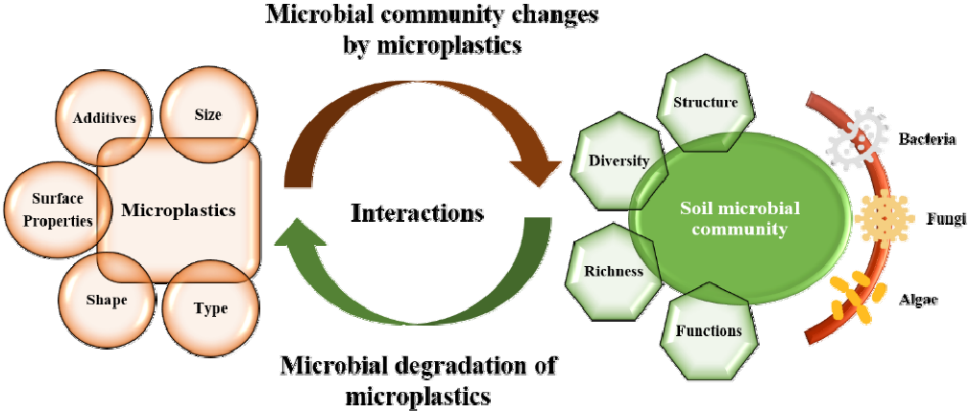
Received November 29, 2022 **Revised** February 2, 2023 **Accepted** February 19, 2023

ABSTRACT

The mass production, continual usage, and improper disposal of plastic products have resulted in significant environmental pollution. The larger plastic polymers gradually break down into smaller particles called microplastics (<5 mm). Existing studies on the occurrence and ecological impact of microplastics have focused on the aquatic ecosystems, with very little attention given to the soil environment. The soil represents a natural sink for microplastics from sources such as sewage sludge, landfills, plastic mulch from agricultural activities, fertilizers, and municipal wastewater effluent. The current study, therefore, provides an overview of existing knowledge on soil microplastic pollution focusing on the impact of microplastics on soil microbial community and microbial degradation of microplastics in soil to systematically identify knowledge gaps to be filled with further research. Future research challenges to be addressed include detailed monitoring of the sources and distribution of microplastics in soil under different land uses, exploring diverse microorganisms in their natural environments for their microplastic biodegradation potential using cultivation-dependent and independent approaches, understanding the mechanism of ecological impacts of microplastics and contributions of microplastic additives, degradation products, and other adsorbed environmental pollutants on soil microbial community.

Keywords: Biodegradation, Microorganism-microplastic interactions, Microplastics, Microplastic additives, Soil microbial community

Graphical Abstract



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1. Introduction

Due to their durability, lightweight, and cost-effectiveness, plastics are widely and frequently used synthetic materials [1]. Almost every aspect of human activities has found synthetic plastics useful, and it can be concluded that, in recent years, plastic materials have grown to become indispensable, replacing other materials such as wood, metals, and glass in various applications. Different synthetic plastics have been used in the production of various materials such as polyethylene (PE) for shopping bags, plastic bottles, and toiletry bottles; polystyrene (PS) for food containers, packaging foams, and disposable cups; polyurethane (PU) for sealants, adhesives, and extrusion and injection-molded parts; polyvinyl chloride (PVC) for plumbing pipes and guttering, window frames, and shower curtains; polypropylene (PP) for microwavable containers, drinking straws, and plastic pressure pipe systems; polyethylene terephthalate (PET) for plastic films, engineering components, and carbonated drinks bottles; nylon for gears, bushings, and plastic bearings; polycarbonate for automobile components, plastic lenses, riot shields; and polytetrafluoroethylene (PTFE) for hookup wire, coaxial cables, and gaskets [2].

Since 1950, the production of plastics has rapidly increased on a global scale due to their extensive use. Between 1950 and 2019, the annual global production increased dramatically from 2 million tons to 368 million tons, with about 40% going toward single-use applications [3]. Virgin plastics produced are largely single-use convenience products that are discarded within a short period after use, and they result in a rapid and massive accumulation in the natural environments [4]. It is predicted that, by 2050, up to 26 billion tons of plastic waste will be generated and more than 50% will be discarded into landfills and eventually enter natural environments like oceans, lakes, rivers, cultivated lands, etc., thereby resulting in serious environmental pollution [5]. Undoubtedly, as a result of its wide distribution across the world [6-8], plastic pollution has become a global scale issue, and the fate of plastic waste in the environment is now a subject of increasing concern.

The term “microplastic (MP)” is proposed to be first used by the African scientists named Ryan and Moloney in 1990 in their research article titled “Plastic and other artefacts on South African beaches: temporal trends in abundance and composition” [9]. This term, however, became widely recognized among researchers after a report published by Thompson et al. [10] who examined the abundance of microplastics (MPs) in the sediment of beaches, estuarine, and subtidal around Plymouth in the United Kingdom. Since then, the term “microplastics” has been generally used to describe small particles of plastics [11]. The defining characteristics of MPs are still under debate [12], but it is agreed by most researchers that plastic particles ranging between 100 nm to <5 mm in size are regarded as MPs.

In the past years, most studies on MP pollution have focused on the marine environment, with very little attention drawn to the terrestrial environment, although between 2018 and 2021, more researchers have studied the effect of MPs on the soil ecosystem [13, 14]. Due to the critical role that the soil plays in regulating nutrient cycling, maintaining the biodiversity of organisms, and providing food [15], it is necessary to evaluate the ecological effect of MP pollution in the terrestrial environment, especially the soil [14, 16]. After the findings of Rillig [17] on the detrimental effects of MPs in soil and terrestrial ecosystems, more research interests have been drawn to plastic pollution in soil. Studies have shown that the soil environment receives much more plastic waste than the marine environment [18] and researchers have warned about the ecological effects of plastics and small plastic particles in soil and terrestrial environments [17, 19]. The main effects of MPs in soil are likely to occur at the interface between soil particles and plastic polymers (i.e., plastisphere). Similar to interactions in the plant rhizosphere, the physicochemical properties of MPs could stimulate the diversity and activities of soil microbial communities at the soil-plastic interface. These interactions can favor the proliferation and activities of specific microbial taxa, and lead to the formation of microbial hotspots in the soil [20]. With the increasing rate of MP contamination in most agricultural soils [21], the specific microbial niches at the soil-MP interface (i.e., microplastisphere) are of ecological importance. However, the effect of MPs on soil microbial communities and its corresponding impact on biodegradation in soil remains largely unclear.

Because of their physicochemical properties, which enhance their resistance to degradation, plastics can accumulate in natural ecosystems. High crystallinity, high molecular weight, and absence of functional groups that favor oxidative reaction processes are contributing factors to the non-biodegradability of plastics [22]. Current methods for managing plastic waste including incineration, landfilling, and recycling have associated demerits that could lead to further environmental issues. For instance, more toxic and volatile waste materials such as nitrogen oxides, furans, heavy metals, sulfides, and dioxins, which are considered to have potential carcinogenic effects, are produced when different synthetic plastics are incinerated [23]. Also, cost-ineffectiveness [24] and down-cycling [25] are undesirable consequences associated with recycling synthetic plastics. Landfilling, a widely used method for plastic waste disposal, especially in developing countries, results in a huge accumulation of plastic waste occupying a vast amount of land. Due to these, efforts are recently being made by researchers to explore other environmentally friendly and sustainable approaches to manage plastic waste and decrease environmental pollution caused by plastic waste. Microorganisms have been explored and reported as promising alternative for the degradation MPs. A number of research reports have identified different microbial species capable of degrading MPs. Although most reports available focused mainly on the biodegradation of a single kind of plastic such as PET [26], PE [27], PU [28], PS [29], and PP [30]. More recently, insects in their larva form including waxworm, mealworm, and superworm have also demonstrated the ability to eat, degrade, and mineralize various MPs, though not without the support of the microorganisms residing in their guts [31]. Research ideas focusing on the biodegradation of all main types of MPs are necessary as well as the biological upcycling of plastic waste [32].

Therefore, the aim of this study is to review previous literatures on the impact of MPs on soil microbial communities and biodegradation of MPs in soil. Also, extensively described are the

Table 1. Response of Soil Microbial Communities to Different Plastics

Soil type	MP type[a]	MP form and concentration[b]	Enriched microbial taxa	Reference
Bacteria				
Field soil	Plastic mulching	n.a.	Cyanobacteria	[49]
Cinnamon soil	LDPE	Fragments, 2000 fragments (kg of soil)$^{-1}$	Actinobacteria, Bacteroidetes, Gemmatimonadetes, and Proteobacteria	[51]
Loamy soil	LDPE, PVC	Particles, 1% and 5% (w/w)	Proteobacteria	[61]
River shore soil	PE, PP, PA, PS, PET, PVC	Particles, 2% (w/w)	Actinobacteria	[62]
Farmland loamy soil	HDPE, PLA	Particles, 0.5% and 10% (w/w)	Actinobacteria, Proteobacteria, Chloroflexi, Gemmatimonadetes	[63]
Rice paddy soil	PS, PTFE	Particles, 0.25% and 0.5%	Chloroflexi and Acidobacteria	[64]
Farmland soil	PE, PS, PVC	Particles, 7% and 14% (w/w)	Proteobacteria and Actinobacteria	[65]
Tea garden soil	PVC	Particles, 900 mg (3.5 kg soil)$^{-1}$	Proteobacteria and Firmicutes	[66]
Field soil	Plastic film	Film, n.a.	Proteobacteria, Bacteroidetes, and Cyanobacteria	[67]
Field soil	PE	Film, 5% (w/w)	Proteobacteria, Actinobacteria, and Acidobacteria	[68]
Fungi				
Field soil	Plastic mulching	n.a.	Ascomycota and Basidomycota	[49]
Farmland soil	PE, PS, PVC	Particles, 7% and 14% (w/w)	Ascomycota	[65]
Field soil	PE	Film, 5% (w/w)	Ascomycota	[68]
Grassland soil	Plastic fragments	Fragments, n.a.	Mortierellomycota and Ascomycota	[69]
Compost	PE, PVC, PHA	Particles, 0.5% (w/w)	Ascomycota and Basidomycota	[70]

[a]PE: Polyethylene; PVC: Polyvinyl chloride; PS: Polystyrene; PP: Polypropylene; PET: Polyethylene terephthalate; HDPE: High-density polyethylene; LDPE: Low-density polyethylene; PLA: Polylactic acid; PA: Polyamide; PTFE: Polytetrafluorethylene; PHA: Polyhydroxyalkanoates
[b]n.a. information not available

current knowledge and knowledge gaps regarding the interactions between MPs and microbial communities in soil. Additionally, an effort has been made to outline the MP-related factors that affect the structural composition, diversity, and functionality of soil microbial communities. Finally, the challenges that need to be addressed in order to fill the knowledge gaps are itemized, along with future research prospects.

2. Sources of MP Pollution in the Natural Environment

Based on source points and their formation pathway, MPs in the environment can be categorized into two, namely; primary MPs and secondary MPs [8]. Primary MPs are synthesized from manufacturing activities and designed for commercial purposes, such as manufacture of personal care and cosmetic products (e.g., microbeads in hand and facial cleansers, toothpaste, shower gel), appliance manufacturing, industrial abrasives (e.g., air blasting), and textile fibers in clothing (e.g., acrylic fibers) [8, 33]. Primary MPs can be produced from the air-blasting industry as a result of the abrasion of materials during the process of preproduction of resin pellets [34]. Production of secondary MPs, on the other hand, is from physical (e.g., wave strike, abrasion, and water disturbance), chemical (e.g., UV radiation, the freeze-thaw cycle), and biological (e.g., biodegradation) activities, which involve the degradation and fragmentation of larger plastic materials into micro-sized particles [34].

The sources of MPs in the soil include vinyl mulch commonly used in agricultural activities [35], domestic sewage water containing MP beads from biosolids, personal care products, and fibers from clothing materials [36], landfills from industrial and urban centers [37], fertilizers [18], illegal waste dumping, irrigation with wastewater, littering roads and lake water flooding [38], atmospheric MP particles transported over long distances [39], and tire abrasion [40]. These MP particles settle on the soil surface and can penetrate into subsoils via physical and environmental activities. Recently, increasing attention has been drawn to MP pollution in the soil ecosystem from various pathways [41]. Studies have not revealed the transfer or presence of MPs in groundwater; however, there is growing concern over the potential distribution and transportation of MPs into the groundwater system and the

hyporheic zone based on previous investigations on the mechanism of transport of MPs [38].

As more attention is drawn to MP pollution in the soil environment, it is necessary to understand the interaction and response of soil microbial communities to MPs in the soil and its effect on MP biodegradation. The process of microbial degradation of MPs in soil is governed by the diverse soil microbial communities that are able to colonize the MP surface, form biofilms, and establish a microenvironment that facilitates MP degradation [42] in the soil environment.

3. Microplastic Pollution and Soil Microbial Community

3.1. Microbial Interactions with MPs in Soil

Microorganisms interact with MPs in different environments, including the soil [43]. As more MPs are introduced in the soil, there is an increasing concern about their ecological effect, especially their effect on soil microbiota. For example, the worldwide use of plastic mulch films in agricultural activities increased from around 4.4 million tons in 2012 to about 7.4 million tons in 2019 [44], and this represents a significant source of MP contamination in the terrestrial environment [45]. Investigations have revealed that several groups of microorganisms including bacteria, fungi, and algae are found attached to MP surfaces [46, 47]. Previous studies have shown that different types of MPs can selectively stimulate the proliferation of specific soil microbial taxa in a microbial community (Table 1). For example, specific soil bacterial and fungal taxa were enriched when exposed to different plastic types (Table 1). It is interesting to note that, in the soil environment, the microbial richness and diversity on MP surfaces are unique and less diverse than those found colonizing natural materials such as wood [48] and the surrounding rhizospheric soil [49]. Due to their uniqueness, the microbial communities on MPs have been referred to as microplastisphere/plastisphere [50].

A few studies have investigated the effect of MPs on the composition and diversity of soil microbial communities; however, their reports remain inconsistent. While some studies have reported that no significant effect was observed on soil microbial community structure [51-53], others reported considerable changes in the abundance and diversity of soil microbial communities exerted by MPs which affect the overall soil function [54]. Yi et al. [55] found that the alpha diversity and soil microbial communities changed significantly with membrane-like PE and fibrous PP, and this resulted in the abundance of Acidobacteria and Bacteroidetes, while Deinococcus-Thermus and Chloroflexi decreased in abundance. In another report, the substrate-induced respiration rate was reduced and significant changes in the root colonization rate of arbuscular mycorrhizal fungi were observed with the addition of MPs, and these suggest that the presence of MPs caused changes in microbial functions [53].

The soil treated with 0.007% low-density polyethylene (LDPE) resulted in three times higher specie turnover rate of bacterial communities than the untreated soil, and the divergence of soil microbial communities increased continually as the LDPE exposure time prolonged [56]. Ng et al. [57] also observed that the addition of LDPE (3% w/w) and PET (0.2% and 0.4% w/w) to the growth medium affected the even distribution and richness of soil microorganisms and soil microbial functions, and 14 unique bacterial genera were predominant and enriched. Liu et al. [58] reported that PP (7% and 28%) had a positive effect on the activities of soil microbiota, while de Souza Machado et al. [59] and Awet et al. [60] observed that polyester (0.05-0.4%), polyacrylic (0.05-0.4%) and PS (1 mg kg^{-1}) showed significant negative effects on soil microbial activities. From these studies, it is quite difficult to draw a general conclusion on the effect of MPs on soil microbial communities as the types, sizes, shapes, and concentrations of MPs, as well as environmental conditions varied in these investigations. The different physical, chemical, and biological properties of the soil types used in these investigations might also have played a role in their findings. However, further research is required to gain deeper insight and validate the effect of co-interaction of soil properties and MP properties on soil microbial communities.

3.2. Factors Influencing the Interaction and Response of Soil Microbial Community to MPs

The structure, diversity, and richness of soil microbial communities can be significantly impacted by MP-related properties. The size of MPs might influence the microbial attachment and colonization as well as the distribution and diversity of soil microbial communities, although existing information on this remains scarce. For example, LDPE powder (150-250 μm) at 2% and 7% (w/w) showed differential tolerance on the bacterial diversity at the genus level [71], but LDPE particles (30 μm) at 0.2% (w/w) had no effect on the microbial community [72]. These findings corroborated the earlier report of Frère et al. [73] who found that MP size, regardless of the MP type, had no significant effect on the composition and diversity of microbial communities attached to them. These, however, contradict the report of Guo et al. [74] who found a significant increase in the richness of soil microbial communities in the presence of PE microfiber of <2 mm long. It was also observed that Actinobacteria was significantly enriched on PE, PS, and PP MPs when compared with their macroplastic forms [75]. These findings indicate that the MP size might play a role in microbial diversity and richness; however, more investigations are needed for a better understanding of the effect of MP size on soil microbial communities.

Apart from the size of MPs, the type of MPs could affect the composition, richness, and diversity of soil microbial communities. The soil samples treated with PE, PS, and PP of the same size (150 μm) and concentration (1% w/w) showed that the microbial communities of the soils treated with PP and PE responded differently from the PS-treated soil, although no major effect was observed in the bacterial diversity among the different MP types [76]. Similarly, the treatment of soil with LDPE particles (150-250 μm) and PS particles (0.33-0.64 μm) did not show significant changes in bacterial diversity [71, 77], whereas the treatment of soil with LDPE particles (678 μm) and PVC particles (15 μm) of the same concentration resulted in a decrease in the Shannon diversity

indices of microbial communities [61]. Also, the increases in the relative abundances of Bacteroidetes and Gemmatimonadetes in the PP- and PE-treated soils were greater than that observed in the PS-treated soil [55]. The disparities in these findings can be attributed to the vast and robust microbial diversity in different soils with varying capacities to respond to disturbances [78] and responses of soil microorganisms to single artificial pressure are not always straightforward [79]. The hydrophobicity and roughness of MP surfaces are important parameters that could influence the microbial attachment and colonization of MPs and the resulting microbial communities [80]. Microorganisms colonizing MP surfaces are found to secret several extracellular polymeric substances (EPS), which act as bio-adhesives to enhance their attachment to MP surfaces [80]. Information on the relationship between MP surface hydrophobicity and soil microbial community and their interaction is still limited. However, it was reported that during the early colonization stage, marine bacteria adhere to the hydrophilic carrier interface for biofilm formation [81]. It was observed that hydrophilic groups such as C-O and C=O increased on PE surface in seawater, leading to a significant decrease in the MP hydrophobic properties [82].

As biofilms form and mature on MP surfaces, the bacterial hydrophobicity positively enhances the adhesion of the microbial communities to the MPs [83]. During the MP aging process, MP properties such as surface topography, polarity, surface area, and roughness could change [84]. These physical and structural changes could exert a direct influence on the composition and diversity of the associated microbial communities. For example, Betaproteobacteria was dominant on the smooth PS surface, while Gammaproteobacteria was dominant on the rough PE surface [85]. A rougher MP surface was found to have a positive correlation with the growth rate and density of the adhered microorganisms [83]. During the early biofilm colonization stage, *Vibrio crassostreae* colonized the smooth PS surface faster (<10 h) than the rough PS surface (6 d); however, rapid decolonization was also observed with the smooth PS surface (i.e., zero after 24 h) [86]. Furthermore, the surface charge on MPs plays a significant role on the abundance and diversity of microbial communities; plastic surface energy of 31–43 mN m^{-1} was discovered to be ideal for microbial colonization [87].

Plastic additives such as citrate esters, phthalate esters, fatty acid esters, glycerides, and polyhydric alcohols have the potential to leach out during the plastic weathering process [88], and their concentrations might increase in soil over time and cause significant changes to the abundance, diversity, and activities of soil microbial communities. Few studies have investigated the effect of plastic additives on soil microbial community and microcosm activity [89-91]. For instance, soil treated with diethyl phthalate (DEP) and di (2-ethyl hexyl) phthalate (DEHP) at 0.1 mg g^{-1} had no significant effect on the structural and functional diversity of the soil microbial community [91]. Also, the increasing dibutyl phthalate (DBP) concentration in soil decreased the bacterial diversity and the relative structure, composition, and abundance of soil bacterial communities reflected a successional change as DBP undergoes degradation [90]. Although plastic additives are not often observed to have a profound impact on soil microbiomes at concentrations of environmental relevance [91], more studies on the impact of different categories of plastic additives on soil microbial communities could throw more insight into understanding the microbial colonization pattern of MPs in the natural environment.

Other factors beyond MP-related properties could also play significant roles in the structural composition, diversity, and activities of soil microbial communities. Exposure duration of soil microorganisms to MPs could impact the composition and diversity of the microbial communities associated with MPs [92, 93]. Molecules adsorbed to MPs such as sugars, nucleic acids, proteins, fatty acids, and lipids have also been shown to improve the primary colonization process of microorganisms [94]. Environmental conditions such as nutrient availability (organic and inorganic nutrients), presence/absence of other pollutants, temperature, pH, light intensity, ionic strength, salinity, and biotic factors are critical to the composition, richness, diversity, and activities of microbial communities associated with MPs [48, 95]. Similarly, seasonal variations and geographical locations have been demonstrated to have significant influence on the diversity of microbial communities associated with MPs [95, 96]. So far, the mechanism of colonization of microbial communities on MPs is still largely unknown. When plastic is introduced to the soil, a series of physicochemical changes occur as a result of natural weathering activities [97], which could impact the soil microbial communities. Virgin plastics are normally smooth with a uniform structure and nearly no surface charge [98]. When exposed to sunlight, photooxidation occurs where solar photons with wavelengths within the UV and blue spectrum strike chromophores in the plastic polymer to initiate photooxidative decomposition [97]. The photooxidative weathering process creates radicals that are responsible for chain scissions in the plastic polymer and their reaction with oxygen oxidizes the polymer surface. This results in an increase in hydrophilic groups [30] and carbonyl groups that enhance the primary attachment and colonization of microorganisms on plastic surfaces for microbial community formation and biodegradation [87, 99, 100]. Plastic additives such as plasticizers, UV stabilizers, heat stabilizers, flame retardants, and pigment agents are also partially degraded during photooxidation [97] and might play a critical role in shaping the composition, abundance, and diversity of microbial communities associated with MPs in the soil [52, 99]. Similar to photooxidation, thermal weathering processes also generate radicals through chain scissions at high temperatures, which enhance microbial colonization and biofilm formation on MP surfaces, and biodegradation [101]. Also, MP fragmentation by mechanical stresses in the soil and biogeochemical attacks reduce the hydrophobicity, brittleness, and stiffness of MPs, thereby, enhancing microbial colonization and the formation of plastic-associated microbial communities [27, 97].

4. Microbial Degradation of MPs

Generally, synthetic plastics can resist degradation and persist in the environment for a long period of time. In the natural environment, the degradation of MPs is an integrated process that in-

Table 2. Plastic Degradation by Actinomycetes, Biodegradation Assay Conditions, and Applied Detection Methods

Actinomycetes strain	Plastic type[a]	Biodegradation assay conditions	Biodegradation detection method	Polymer reduction (%)[b]	Reference
Streptomyces scabies	PET	Ten (10) mg of PET (ground granules), 1 mL of Tris-HCl (20 mM, pH 7.5), and 3 μg of the enzyme Sub 1 from *S. scabies*. Incubation for 15 d at 37℃	Measurement of the amount of terephthalic acid (TA) released from PET	n.a.	[112]
Rhodococcus ruber C208	PE	Synthetic media, for 2 months, 150 rpm, at 30℃	Weight loss; SEM	7.5	[116]
Streptomyces sp.	PET	Mineral salt media, for 18 d, 120 rpm at 28℃	Weight loss, SEM	68.8	[117]
Rhodococcus rhodochrous ATCC 29672	PE	Mineral salt media, for 6 months, at 27℃ and 85% humidity	FTIR; SEM; GPC	n.a.	[118]
Micrococcus sp.	PE	Nutrient broth media, for 1 month	Weight loss	6.61	[119]
Bacterial consortia (*Arthrobacter viscosus; Micrococcus lylae; M. luteus; Bacillus mycoides; B. cereus; B. pumilus; B. thuringiensis*)	PE	Films buried in soil for 7.5 months, at room temperature	Weight loss; FTIR; SEM; elongation at break	17.0	[120]
Streptomyces sp.	HDPE	Mineral salt media, for 18 d, 120 rpm at 28℃	Weight loss; SEM	18.26	[121]
Microbacterium paraoxydans	PE (pre-treated with nitric acid)	Minimal broth media, for 2 months, 180 rpm, at room temperature	Weight loss; FTIR	61.0	[122]
Streptomyces coelicoflavus NBRC 15399T	LDPE	Mineral salt medium for 4 weeks	Weight loss; pH change, trinocular microscope	30	[123]
Streptomyces badius	Starch-PE (10 d-heat-treated)	0.6% yeast extract media, for 0.75 months, 125 rpm, at 37℃	FTIR; tensile strength at break; GPC	n.a.	[124]
Streptomyces setonii	Starch-PE (10 d-heat-treated)	0.6% yeast extract media, for 0.75 months, 125 rpm, at 37℃	FTIR; tensile strength at break; GPC	n.a.	[124]
Streptomyces viridosporus	Starch-PE (10 d-heat-treated)	0.6% yeast extract media, for 0.75 months, 125 rpm, at 37℃	FTIR; tensile strength at break; GPC	n.a.	[124]
Streptomyces sp.	LDPE	M1 media for 45 d, 120 rpm at 25℃	Weight loss; Sturm test, AFM, FTIR	5.2	[125]
Rhodococcus ruber	PS	Synthetic media, for 2 months,	Weight loss; SEM	0.8	[126]
Rhodococcus sp. strain 36	PP	Bushnell Haas (BH) media, for 1.25 months, at 29℃	Weight loss; FTIR; SEM	6.4	[114]
Rhodococcus rhodochrous ATCC 29672	PP (pre-treated with photo and thermo-oxidation)	Mineral media, for 6 months, at 27℃	FTIR; 1H NMR; ADP/ATP ratio	n.a.	[127]
Streptomyces sp.	PE	Mineral salt medium for 6 months, 150 rpm at 30℃	Clear zone test; weight loss.	46.16	[128]
Streptomyces sp.	Plastic bag	Mineral salt medium for 6 months, 150 rpm at 30℃	Clear zone test; weight loss.	35.78	[128]

Actinomycetes strain	Plastic type[a]	Biodegradation assay conditions	Biodegradation detection method	Polymer reduction (%)[b]	Reference
Micrococcus sp. AF10	PU	Films buried in soil, for 6 months, at 30-35℃	Clear zone test; SEM; FTIR	n.a.	[129]
Arthrobacter sp. AF11	PU	Films buried in soil, for 6 months, at 30-35℃	Clear zone test; SEM; FTIR	n.a.	[129]
Streptomyces spp.	PE (containing 6% starch)	Mineral salt medium containing 0.6% yeast extract for 4 weeks, 125 rpm at 30℃	Weight loss; tensile strength; percent elongation	Inconclusive	[130]
Amycolatopsis orientalis (enzyme production)	PLA (production of purified enzyme)	Incubation for 8h, 140 rpm, at 30℃	FTIR, pH variation; enzyme degrading activity	n.a.	[131]
Saccharothrix waywayandensis	PLA	Basal media with 0.1% gelatin, for 0.25 months, 180 rpm, at 30℃	Weight loss; pH variation	95	[132]
Kibdelosporangium aridum	PLA	Basal media with 0.1% gelatin, for 0.5 months, 180 rpm, at 30℃	Weight loss; pH variation; SEM	97	[133]
Amycolatopsis sp.	PLA	Basal medium for 14 d, 180 rpm at 30℃	Weight loss; plate count; clear zone method	60	[134]
Actinomadura sp. T16-1 (enzyme production)	PLA (production of PLA-degrading enzyme)	Basal media with 0.2% gelatin, for 96h, 150 rpm, at 50℃	Enzyme activity	n.a.	[135]

[a]PE: Polyethylene; PET: Polyethylene terephthalate; HDPE: High-density polyethylene; LDPE: Low-density polyethylene; PS: Polystyrene; PP: Polypropylene; PU: Polyurethane; PLA: Polylactic acid; SEM: Scanning electron microscope; FTIR: Fourier-transform infrared spectroscopy; GPC: Gel permeation chromatography; AFM: Atomic force microscopy; NMR: Nuclear magnetic resonance spectroscopy; ADP: Adenosine diphosphate; ATP: Adenosine triphosphate
[b]n.a. information not available.

corporates physical, chemical, and biological elements [102]. Despite MPs being less susceptible to degradation as compared to other biodegradable materials, a number of microbial strains have been identified with the potential to biodegrade MPs in the natural environment, including soils of plastic-dumping sites, waste of mulch films, marine water, crude oil-contaminated soils, landfills, sewage sludge, and guts of plastic-eating insects [103]. Microorganisms are found everywhere in nature, and they are able to adapt to a variety of environmental conditions, including extreme conditions. Their complex enzymatic system, ability to utilize various organic and inorganic substrates for growth, and ability to adapt to extreme environmental conditions are factors that contribute to their capacity to break down a wide variety of environmental contaminants. Therefore, they represent a sustainable and eco-friendly alternative to the management of plastic pollution in the environment. MPs form a novel ecological niche for microorganisms by providing a support system for colonization and growth, and a source of nutrient for microbial nutritional needs. The attachment of different groups of microorganisms (e.g. bacteria, actinomycetes, fungi, protists, algae, and viruses) to MP surfaces provides them an enabling environment to form biofilms [104]. The result of the microbial enzymatic activities leads to the structural deformation and loss of properties of the MPs [105]. Different categories of microorganisms capable of degrading MPs have been isolated from various environments [106]; however, there is still a knowledge gap about microbial interactions with MPs, which has contributed to the lesser adoption of plastic biodegradation strategies [107]. There are four stages involved in biofilm formation and biofilm-mediated MP degradation [104], and these include 1) microorganism adhesion to MP surface and modification of their surface properties, 2) MP deterioration, in which microbial enzymes speed up degradation and liberate monomers and additives from the MPs, 3) MP fragmentation, where the MPs lose their mechanical stability and become fragile as a result of microbial attack, and 4) assimilation of MPs, which involves the penetration of microbial filaments and water and the subsequent microbial decomposition and use of MPs as a nutrient source.

4.1. Actinomycetes in MPs Biodegradation

Actinomycetes, a group of gram-positive, filamentous bacteria, can be found in various ecological habitats including soil, plant tissues, freshwater, and marine environments [108]. Actinomycetes are recognized for their metabolic versatility and can perform different functions in the environment. They are explored for numerous biotechnological applications such as the production of enzymes,

Table 3. Plastic degradation by bacteria, biodegradation assay conditions, and applied detection methods

Bacterial strain	Plastic type[a]	Biodegradation assay conditions	Biodegradation detection method	Polymer reduction (%)[b]	Reference
Anoxybacillus rupiensis	Nylon 6	Chemically defined medium containing 0.5% nylon 6 for 7 d, 180 rpm at 65℃	HPLC; FTIR	n.a	[144]
Bacillus cereus	PE; PET; PS; pre-treated with UV	Mineral salt media with 0.5 g polymer, for 1.25 months, 150 rpm, at room temperature	Weight loss; FTIR; SEM	1.6; 6.6; 7.4, respectively	[137]
Bacillus gottheilli	PE; PP; PET; PS; pre-treated with UV	Mineral salt media with 0.5 g polymer, for 1.25 months, 150 rpm, at room temperature	Weight loss; FTIR; SEM	6.2; 3.0; 3.6; 5.8, respectively	[137]
Achromobacter denitrificans	PE pre-treated with UV and heat (70℃ for 3 d)	Synthetic nutrient medium with 0.1 g polymer for 2 months, 180 rpm at 30℃	Weight loss; tensile strength; NMR; XRD; thermo-gravimetric analysis; carbon analysis; GCMS	40	[145]
Bacillus sp. YP1	PE	Liquid carbon-free media (LCFBM) with 1 g polymer, for 2 months, 120 rpm, at 30℃	Weight loss; FTIR; SEM; GPC; tensile strength	10.7	[146]
Enterobacter asburiae YT1	PE	LCFBM with 1 g polymer, for 2 months, 120 rpm, at 30℃	Weight loss; FTIR; SEM; GPC; tensile strength	6.1	[146]
Acinetobacter baumannii	PE	Synthetic medium with film for 30 d, 100 rpm at 37℃	FTIR; GC-MS; tensile strength	n.a	[147]
Indigenous Marine Microbial Community bioaugmented with *Lysinibacillus* sp. and *Salinibacterium* sp.	PE	Nutrient broth media enriched with saline water, for 6 months, 120 rpm, at 25℃	Weight loss; FTIR; SEM	19	[148]
Bacillus cereus	PE	Basal medium with polymer strip for 2 months	GC-MS	n.a	[149]
Mesophilic mixed Bacterial Culture (*Bacillus* sp. and *Paenibacillus* sp.)	PE	Basal media with 100 mg of polymer, for 2 months, at 30℃	Weight loss; FTIR; SEM	14.7	[106]
Bacillus spp.	PE	Synthetic media with polymer powder for 16 weeks, 150 rpm at 37℃	Weight loss; FTIR; GC-MS	35.72	[139]
Biofilm composed of *Pirellulaceae, Phycisphaerales, Cyclobacteriaceae,* and *Roseococcus*	PE and PP	Dechlorinated tap water, Woods Hole media, for 0.7 months (incubation in tanks in a greenhouse exposed to natural light)	DNA extraction, amplification and sequencing (evaluation of the effects of substrate type on microbial communities)	n.a	[105]
Bacillus amyloliquefaciens	PE	Bushell Haas media with polymer film for 2 months, 120 rpm at 28℃	Weight loss; GPC; FTIR; DSC; TGA; ESI-MS	3.2	[150]
Brevibacillus borstelensis	PE	Synthetic media with polymer powder for 16 weeks, 150 rpm at 37℃	Weight loss; FTIR; GC-MS	20.28	[139]
Bacillus sp. strain 27	PP	Bushell Haas media with 0.5 g polymer, for 1.25 months, at room temperature	Weight loss; FTIR; SEM; pH variation	4	[114]
Pseudomonas sp.	PS	Mineral salt media with polymer beads for 1 month, 120 rpm at 37℃	GC-MS	na	[151]
Pseudomonas aeruginosa	PU	Trypticase soy broth media, for 72 h, at 37℃	Weight loss; SEM; tensile strength and elongation at break	2.5	[152]

Bacterial strain	Plastic type[a]	Biodegradation assay conditions	Biodegradation detection method	Polymer reduction (%)[b]	Reference
Escherichia coli	PU	Trypticase soy broth media, for 72 h, at 37°C	Weight loss; SEM; tensile strength and elongation at break	2.4	[152]
Pseudomonas aeruginosa	PS-PLA Nanocomposites	Minimum salt media, for 0.94 months, at room temperature	Weight loss; FTIR; SEM; pH variation	9.9	[153]
Stenotrophomonas maltophilia LB 2-3	PLA	Mineral medium, for 1.33 months, at 37°C	GPC; FTIR; NMR; tensile strength	n.a	[154]

[a]PE: Polyethylene; PET: Polyethylene terephthalate; PS: Polystyrene; PP: Polypropylene; PU: Polyurethane; PLA: Polylactic acid; HPLC: High-performance liquid chromatography; FTIR: Fourier-transform infrared spectroscopy; SEM: Scanning electron microscope; NMR: Nuclear magnetic resonance spectroscopy; XRD: X-Ray diffraction analysis; GC-MS: Gas chromatography–mass spectrometry; GPC: Gel permeation chromatography; DSC: Differential scanning calorimetry; TGA: Thermogravimetric analysis; ESI-MS: Electrospray ionization mass spectrometry, [b]n.a. information not available

anticancer drugs, antibiotics, and other bioactive metabolites [109]. Some strains of actinomycetes have also been used for the bioremediation of toxic materials, breakdown of resistant carbohydrates (e.g. chitin, cellulose), and recycling of organic carbons [110]. Their ability to produce hydrolytic enzymes and bioactive metabolites enables them to grow on and degrade many complex polymers [111] including MPs [112]. Actinomycetes are known to produce extracellular biopolymers like levan, glycogen, dextran, and *N*-acetylglucosamine-rich slime polysaccharides, which are assumed to enhance their attachment to MP surfaces for subsequent microbial activities [113]. Actinomycetes such as the genera *Streptomyces*, *Actinomadura*, *Rhodococcus,* and thermophilic species of *Thermoactinomycetes* have been isolated from different environments with significant plastic degradation potential [112, 114]. Other actinomycetes species that have been reported to be associated with MP degradation are listed in Table 2. From the list, it is evident that limited actinomycetes genera have so far been identified to have MP-degradation potential, predominantly members of *Streptomyces*, *Rhodococcus*, and *Micrococcus*. Reports have also shown that some actinomycetes can form biofilm, similar to other bacterial strains, which is important for their survival and colonization on MPs [115].

4.2. Other Bacteria in MPs Biodegradation

Bacteria are the most abundant group of microbes. They are well recognized for their ubiquity and can be found in various environments such as water, soil, and atmosphere. Numerous bacterial species have been extensively studied for their roles in the biodegradation of complex polymers and bioremediation of environmental pollutants such as metal compounds, crude oil, antibiotics, plastic, and other compounds of environmental concern [136]. With their diversity and metabolic activities, bacterial strains are able to adsorb, desorb, and break down MPs [114, 137]. Different bacterial strains have been reported to utilize MPs as their main carbon source in minimal medium, and with their metabolic activities, they induced significant weight loss and changes to the morphological and chemical structure of MPs (Table 3).

Plastic-degrading bacteria have been isolated from different environments such as cold marine environment [138], dumpsites [139], landfills [140], recycling sites [141], and insects' guts [142]. Most studies on the bacterial degradation of MPs have used pure bacterial cultures isolated from different environments or obtained from culture collections for the degradation of MPs under laboratory conditions. The use of pure bacterial strains can be advantageous when investigating specific metabolic pathways, evaluating different conditions, and/or closely monitoring the process of MP degradation [143]. However, in nature, many bacterial strains act in synergy, forming consortium and constituting a stable microbial community, which ultimately enhances their survival and degradation potential. So far, few studies have focused on MP biodegradation by bacterial consortia [129], it is, therefore, essential that more investigations be conducted to explore the potential of different bacterial consortia in MP biodegradation, as this might lead to greater efficiency due to the metabolic synergism between different bacterial strains.

4.3. Algae in MPs Biodegradation

In the last decades, algae have been extensively studied for their biotechnological applications, especially in the production of biofuels [155, 156]. The ability of different photosynthetic and heterotrophic algae to degrade environmental pollutants, both organic and inorganic, has also been well studied and established [157]. They are able to degrade environmental pollutants by adsorbing, accumulating, or metabolizing them into safer levels [156, 157]. Most studies on the environmental biodegradation of MPs have focused on the potentials of other groups of microorganisms such as bacteria and fungi, and only a few studies have investigated the potential of algae for biodegradation of MPs. Rather, recent studies have focused on the potential of algae in green plastic production [158].

Algal species including *Oscillatoria, Spirogyra, Anabaena, Spirulina*, and *Chlorella* have been reported to be found colonizing MP surfaces in terrestrial environments, but it is still inconclusive to confirm that these algal species are able to metabolize the MPs [159, 160]. In a study by Khoironi et al. [161], Spi*rulina* sp. was able to biodegrade PP and PET during 112 d incubation. The authors reported that the tensile strength of PP and PET decreased by 0.1977 MPa d^{-1} and 0.9939 MPa d^{-1}, respectively. Kumar et al. [162] also reported that *Navicula pupula, Scenedesmus dimorphus*, and *Anabaena spiroides* showed degradation potentials on

Table 4. Plastic degradation by fungi, biodegradation assay conditions, and applied detection methods

Fungal strain	Plastic type[a]	Biodegradation assay conditions	Biodegradation detection method	Polymer reduction (%)[b]	Reference
Aspergillus flavus	PE	Sole carbon source medium for 28 d, 150 rpm at 28℃	Weight loss; molecular weight shift; surface smoothness change	3.9025	[31]
Aspergillus flavus VRKPT2	PE	Synthetic media with mineral oil, for 1 month, at 30℃	Weight loss; FTIR; SEM	9.34	[172]
Aspergillus tubingensis VRKPT1	PE	Synthetic media with mineral oil, for 1 month, at 30℃	Weight loss; FTIR; SEM	6.88	[172]
Aspergillus flavus	PE (with 6% starch)	Mineral salt medium containing 3% yeast extract for 4 weeks, 125 rpm at 30℃	Weight loss; tensile strength; percent elongation	1.2	[130]
Mucor rouxii NRRL 1835	PE (with 6% starch)	Mineral salt medium containing 3% yeast extract for 4 weeks, 125 rpm at 30℃	Weight loss; tensile strength; percent elongation	Inconclusive	[130]
Aspergillus oryzae	PE	Synthetic medium with ampicillin, for 4 months, at 28℃ in a shaker incubator	Weight loss; FTIR; GC-MS	36.4	[139]
Penicillium simplicissimum YK	PE	Medium C with 0.5 g polymer, for 3 months, 150 rpm, at 30℃	FTIR; GPC	n.a.	[173]
Zalerion maritimum	PE	Minimum growth media with 0.130 g of polymer, for 0.94 months, 120 rpm, at 25℃	Weight loss; FTIR; NMR	43	[164]
Trichoderma harzianum	PE (UV treated)	Mineral salt medium for 3 months	Weight loss; SEM; FTIR; NMR	40	[174]
Pleurotus ostreatus	PE (120 d exposed to sunlight)	Mineral medium, for 2 months, at 25℃	FTIR; SEM; mechanical properties	n.a.	[172]
Aspergillus clavatus JASK1	PE	M1 media for 90 d, 120 rpm, at 25℃	Weight loss; Strum test; FTIR; SEM; AFM analysis.	35	[175]
Cephalosporium sp. (NCIM 1251)	PS	Mineral salt media, for 2 months, at 120 rpm, at 28℃	Weight Loss; FTIR; SEM; pH variation; gel permeation chromatography	2.17	[176]
Mucor sp. (NCIM 881)	PS	Mineral salt media, for 2 months, at 120 rpm, at 28℃	Weight Loss; FTIR; SEM; pH variation; gel permeation chromatography	1.81	[176]
Pestalotiopsis sp.	PS (Styrofoam)	Malt extract broth, for 1 month, at 25℃	Weight loss; FTIR; SEM	74.43	[177]
Ceriporia sp.	PS (Styrofoam)	Malt extract broth, for 1 month, at 25℃	Weight loss; FTIR; SEM	19.44	[177]
Cymatoderma dendriticum	PS (Styrofoam)	Malt extract broth, for 1 month, at 25℃	Weight loss; FTIR; SEM	15.50	[177]
Cladosporium cladosporioides	PU	Agar medium	Clear zone test; FTIR	n.a.	[178]
Cladosporium pseudocladosporioides	PU	Mineral medium for 14 d at 30℃	Weight loss; FTIR; GC-MS; SEM	87	[179]

Fungal strain	Plastic type[a]	Biodegradation assay conditions	Biodegradation detection method	Polymer reduction (%)[b]	Reference
Phoma sp.	PU	Buried in soil for 5 months	Clear zone test; tensile strength	n.a.	[180]
Aspergillus tubingensis	PU	Mineral salt medium, for 0.75 months, at 150 rpm, at 37℃	FTIR; SEM; tensile strength	n.a.	[181]
Phanerochaete chyrosporium	PVC (blended with cellulose)	Soil buried (soil was mixed with municipal sewage sludge), for 6 months	Clear zone test; FTIR; SEM	n.a.	[182]
Cochliobolus sp.	PVC	Laccase degradation	FTIR; SEM; GC-MS	n.a.	[183]
Tritirachium album	PLA	Basal media, for 0.5 months, at 180 rpm, at 30℃	Weight loss; SEM; pH variation	76	[184]
Thermomyces lanuginosus	PLA	Wheat grain with mineral salt medium, for 2 months, at 50℃	SEM; tensile strength	n.a.	[185]
Trichoderma viride	PLA (plasticized with USE)	Liquid Sabouraud medium, for 0.7 months, at 28℃	Weight loss; FTIR; gel permeation chromatography; SEM	1.2	[186]
Aspergillus nomius	PE	M1 media for 45 d, 120 rpm at 25℃	Weight loss; Sturm test, AFM, FTIR, GC-MS	4.9	[125]

[a]PE: Polyethylene; PS: Polystyrene; PU: Polyurethane; PVC: Polyvinyl chloride; PLA: Polylactic acid; FTIR: Fourier-transform infrared spectroscopy; SEM: Scanning electron microscope; GC-MS: Gas chromatography–mass spectrometry; GPC: Gel permeation chromatography; NMR: Nuclear magnetic resonance spectroscopy; AFM: Atomic force microscopy
[b]n.a. information not available

both LDPE and high-density PE (HDPE). From their findings, *A. spiroides* was able to degrade 8.18% of LDPE after 30 d, representing the most promising algal strain in their study. The reported degradation rate is considerably lower than that of some investigations where bacteria and fungi were used to degrade PE. For instance, *Brevibacillus borstelensis*, a thermophilic bacteria isolated from soil, was able to degrade 30% of PE films after 30 d incubation [163]. A marine fungus, *Zalerion maritumum*, was similarly able to degrade 43% of PE pellets after 28 d incubation [164]. Generally, the low MP degradation rate by algae in comparison with bacteria and fungi is quite understandable. Unlike bacteria and fungi, most algae are considered to be photoautotrophic organisms that use atmospheric CO_2 as their main source of carbon, and their main energy source is derived from sunlight [165]. Some algae are able to grow under heterotrophic conditions where external carbon and energy sources are utilized under dark conditions [166]. Even though they are able to colonize and assimilate MPs, algae are not metabolically inclined to mineralize MPs [162]; therefore, there is the possibility of plastic accumulation in algae which might be introduced to the food chain [167].

4.4. Fungi in MPs Biodegradation

Fungi are known to possess vast metabolic potentials, including the production of extracellular multienzyme complexes [168], thereby making them microorganisms of interest in MP biodegradation research. Different fungal strains can be found in different natural environments and they play significant roles in maintaining biogeochemical cycles and promoting the transformation of different substances [169]. As highlighted in Table 4, different fungal species and consortia have been reported with MP-degrading potential based on their ability to utilize MPs as their sole carbon or energy source, predominantly members of the genus *Aspergillus*, *Penicillium*, *Fusarium*, *Trichoderma*, *Mucor*, and *Cladosporium*. Some edible fungal species such as *Agaricus bisporus, Pleurotus abalones,* and *Pleurotus ostreatus* have also been reported to utilize PE and PS for growth, with changes in laccase activity [170]. Similar to bacteria, fungi are able to adhere to and utilize MPs [171]. Through their metabolic activities, they have the potential to decrease the hydrophobicity of MPs by promoting the formation of chemical bonds like ester, carbonyl, and carboxyl functional groups in MPs [104].

5. Knowledge Gaps and Future Prospects

The majority of research on MP pollution and its ecological effects has focused on the marine environment and other aquatic ecosystems, and there have been limited investigations on MP pollution in soil thus far. Unlike water, the soil is a unique media with relatively complex and distinctive physical, chemical, and biological characteristics; as a result, it is challenging to investigate the ecological effect of MPs, particularly with regard to soil microorganisms, which are crucial to nutrient cycling and maintenance of soil functions. From this study, it is evident that, although research on MPs in soil is increasing as more attention is drawn to this area, there is still a substantial gap in understanding the

interactions of soil microorganisms with MPs as well as the mechanisms by which microorganisms degrade MPs in the soil. The ecological effect of MPs on the structural composition, diversity, and activities of soil microbial communities are far from being fully understood. Therefore, future studies should focus on addressing the following issues. Firstly, it is crucial to monitor the sources and distribution of MPs in the soil. The extent of MP pollution under different land uses and natural environments should be understood. Also, further research is needed to better understand the ecological impact of MPs on soil microbiota and microbial communities. Critical questions to be answered with multiple lines of evidence would include i) what is the mechanism by which MPs affect soil microbial communities?, ii) how do MP properties affect the structural composition, richness, diversity, and activities of soil microbial communities in MP-polluted soils?, iii) would the co-interactions of MP properties and soil properties have a significant impact on soil microbial communities?, iv) do intermediates and products of MP degradation impact the diversity and activities of soil microbial communities?, v) what ecological impact do MP additives and adsorbed environmental pollutants have on the microbial communities in the soil?. Lastly, current literature revealed that information on microbial degradation of MPs has centered on specific groups of microorganisms of limited genera, majorly pure microbial cultures, with a few studies on microbial consortium. This amplifies the necessity to explore the potential of various microbes in their natural environments for MP degradation. As most microorganisms exhibit synergistic interactions in their natural environments, it is suggested that the use of different microbial strains in consortium will result in greater efficiency in MP degradation. Furthermore, the application of omic tools such as metagenomics, genomics, metabolomics, proteomics, and transcriptomics will aid in understanding the biological activities that take place at the genetic and metabolic levels, and the influence of environmental factors during MP biodegradation.

Acknowledgments

This study was funded by the National Research Foundation of Korea (NRF-2021R1A2C4001746).

Conflict of Interest

The authors declare that they have no conflict of interest.

Author contributions

K.C.O. (PhD student) wrote the manuscript. E.H.J. (Associate professor) wrote and revised the manuscript.

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