Evaluación bioelectroquímica preliminar de la degradación de tereftalato de polietileno (PET) por bacterias electrogénicas en celdas de combustible microbianas

Preliminary bioelectrochemical evaluation of the degradation of polyethylene terephthalate (PET) by electrogenic bacteria in microbial fuel cells

Abstract*– The accumulation of polyethylene terephthalate (PET) represents a critical environmental challenge due to its high crystallinity and resistance to natural degradation. This study evaluated the synergistic performance of a bacterial consortium (*Pseudomonas stutzeri*, *Bacillus cereus*, *Stenotrophomonas maltophilia*, and *Actinomyces* sp.) for simultaneous biodegradation of PET and bioelectricity generation in a single-chamber microbial fuel cell (MFC). The consortium (10% v/v, 1×10^8 CFU/mL, 1:1:1:1 ratio) was cultured with 5 g of PET as the sole carbon source in a 500 mL MFC with a carbon-coated copper anode and a zinc cathode, operating under constant agitation (120 rpm) for 98 days. Physicochemical parameters (pH, electrical conductivity, optical density) and electrochemical outputs (voltage, current) were periodically monitored. The system reached a maximum voltage of approximately 1.0 V and a peak current of 0.056 mA. FTIR analysis of the PET samples revealed significant structural changes, including an increase in hydroxyl (-OH) and carbonyl (C=O) groups, and a reduction in the intensity of C-H and C-O-C bonds, suggesting polymer biooxidation and chain cleavage. The evolution of pH (from 7.0 to 9.07) and conductivity (from 2.13 to 8.89 mS/cm) were consistent with microbial activity and biofilm establishment. These findings indicate that the synergistic action of hydrolytic (*Bacillus*, *Actinomyces*) and electrogenic (*Pseudomonas*, *Stenotrophomonas*) bacteria could allow an effective degradation of PET, coupled to a sustained generation of electricity, positioning this MFC system as an opportunity for sustainable entrepreneurship and green innovation for the conversion of plastic waste into value-added energy resources. However, complementary analysis using scanning electron microscopy (SEM) is recommended to confirm the superficial physical damage of the polymer.

Keywords*-- Biodegradation, bioelectricity, microbial fuel cells, PET, bacterial consortium.

I INTRODUCTION

The massive accumulation of plastics represents an unprecedented environmental crisis. In 2019, global production reached 368 million tonnes, of which PET accounted for approximately 30 million tonnes [1]. Despite its ubiquity, the management of this waste is inefficient, as only 21% of total plastics are recycled, which allows an accumulation of 33 billion tons of waste to be projected by 2050 [2,3]. PET is particularly problematic due to its high crystallinity (30-50%), which gives it a shelf life of up to 450 years before degrading naturally [4].

Despite the development of biodegradable plastics, their timely degradation remains a critical challenge [5]. The limitations of traditional recycling methods, including high costs, excessive energy consumption and the generation of toxic by-products, underscore the urgency of seeking sustainable alternatives [6,7]. In this context, Microbial Fuel Cells (MFCs) have emerged as a promising biotechnological alternative by integrating the biodegradation of pollutants with bioelectricity generation.

However, preliminary evaluations report limitations in its performance, with maximum voltages close to 222 mV and PET mass reductions of approximately 30% after 30 days of operation [8]. These results show the need to develop more efficient bioelectrochemical strategies, based on the synergistic action of electrogenic microorganisms and PET-degrading enzymes, which simultaneously improve polymer degradation and energy use, thus contributing to the development of sustainable technologies aligned with the principles of the circular economy [9,10].

The environmental persistence of PET is based on its high structural stability and a degree of crystallinity ranging between 30% and 50%, factors that severely restrict its bioavailability for enzymatic attack [4].

Some studies have documented trials on microbial biodegradation of PET, where enzymes play an important role in its metabolism, for example, the bacterium *Ideonella sakaiensis* secretes PETase enzymes, which hydrolyze PET mainly into mono(2-hydroxyethyl) terephthalate (MHET). Subsequently, the MHETase enzyme breaks down MHET into its monomers: terephthalic acid (TPA) and ethylene glycol (EG) [8,11].

Under anaerobic conditions, *I. sakaiensis* has been found to act as a facultative anaerobe, fermenting these components to produce acetate and ethanol [9]. These metabolites are essential for bioelectrochemical systems, as bacteria such as *Geobacter sulfurreducens* consume the acetate to generate electricity [9]. Evaluations in single-chamber MFCs have achieved a 30% PET mass reduction in 30 days using activated sludge, reaching voltage peaks of 222 mV [8].

Microbial fuel cells are bioelectrochemical systems where microorganisms degrade a substrate through oxidative processes at the anode, releasing electrons that flow through an external circuit to the cathode to generate bioelectricity [12]. In this context, electrogenic bacteria act by transforming chemical energy into electrical energy by transporting electrons to a solid electrode [12]. Extracellular electron transfer occurs primarily by three pathways: direct transfer via membrane cytochromes, transfer mediated by soluble redox compounds (such as flavins), or long-distance transfer via pili

conductors [13]. In the cathode compartment, these electrons combine with protons and an end acceptor, usually oxygen, to form water and close the electrochemical circuit [14]. Species such as *Pseudomonas stutzeri* stand out as bifunctional electrogenic bacteria, using type IV pili to attach to the anode and form electroactive biofilms [15]. Likewise, the genus *Pseudomonas* can produce mediators such as pyoocyanin, which acts as an electron shuttle facilitating transport from cell metabolism to the electrode [16].

First, in [6], Pourhosseini et al. evaluated a bacterial consortium where *P. aeruginosa and Agrobacterium fabrum predominated*, achieving a biodegradation of 21.4% of PET in double-chamber MCC and a power density of 0.343 mW/m² after 60 days of operation. In [17], the use of the fungus *Paecilomyces sp*. in single-chamber MFCs with carbon and zinc electrodes; in this system, a reduction in chemical oxygen demand (COD) of 85.47% and a maximum voltage of 0.575 V were achieved, highlighting that a pH of 6.5 favored the flow of electrons. Finally, operational optimization is crucial for these bacteria; according to [18], strains such as *Pseudomonas sp*. M1 require a neutral pH (6.8 to 7.2) to maximize extracellular electron transfer and the formation of dense biofilms at the anode. Under these optimal conditions, current densities of up to 2.5 mA/cm² were recorded, while deviations towards acidic or alkaline pH reduced electrical production by 30% to 50%. These antecedents confirm that the performance of MFCs depends on the selection of microorganisms with bifunctional capabilities and the optimization of operating factors such as pH, which directly influences enzymatic activity and extracellular electron transfer to the anode.

The scientific gap in this research lies in the poor integration of polyethylene terephthalate (PET) biodegradation and the simultaneous generation of bioelectricity in microbial fuel cells. Although there are isolated studies on the enzymatic degradation or electrogenic performance of certain strains, there is limited evidence evaluating how pH optimization (in a range of 6.0 to 8.0) influences the synergy of a specific bacterial consortium *(Pseudomonas stutzeri, Bacillus cereus, Stenotrophomonas maltophilia* and *Actinomyces sp.*). A lack of knowledge persists about the balance between polymer hydrolysis and extracellular electron transfer in single-chamber systems under these controlled conditions.

Despite the potential of MFCs for waste recovery, a critical gap persists in the integration of electrogenesis with the degradation of polyethylene terephthalate (PET). Currently, no electroactive bacteria with intrinsic capacity to degrade PET directly have been documented. Likewise, research using PET microplastics as the primary substrate in bioelectrochemical systems is scarce. There is also limited understanding of how these polymers affect the viability of anode biofilms and alter long-term electron transfer mechanisms. Finally, the scalability of these processes for industrial application remains an unsolved technical challenge

Therefore, the present research aimed to evaluate the ability of a consortium made up of *P. stutzeri, B. cereus, S. maltophilia* and *Actinomyces sp*. to degrade PET and generate bioelectricity in a microbial fuel cell, by monitoring voltage, current, pH, electrical conductivity, biomass and structural changes of the polymer by FTIR.

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II MATERIAL AND METHOD

2.1 Obtaining, Reactivating and Conditioning Electrogenic Strains

The bacterial strains *Stenotrophomonas maltophilia, Pseudomonas stutzeri, Bacillus cereus and Actinomyces sp*. were provided by a local research institution, having been previously identified using molecular biology techniques [19,20]. For the reactivation and purification process, the species *S. maltophilia, P. stutzeri and B. cereus* were resuspended in nutrient broth and incubated at 35 °C for 24 hours; subsequently, the cultures were seeded by the striation method by exhaustion in plates with nutrient agar and incubated under the same temperature and time conditions. For its part, the reactivation of Actinomyces sp. it was carried out in trypticase soybean broth (TSB) at 30 °C for 5 days, and then sown by depletion in plates with trypticase soybean agar (TSA) and incubated at 30 °C for an additional period of 5 days. Finally, the macroscopic and microscopic purity of the isolated colonies of all strains was confirmed by the Gram staining technique [21].

The conditioning of the Consortium (made up of the four bacterial strains) was carried out by inoculating the microbial group in Bushnell-Haas broth medium. The medium was prepared with the following composition (per liter): $MgSO_4 \cdot 7H_2O$, 0.2 g; $CaCl_2$, 0.02 g; KH_2PO_4, 1.0 g; K_2HPO_4, 1.0 g; NH_4NO_3, 1.0 g; $FeCl_3$, 0.05 g. The pH was adjusted to 7.0 ± 0.2 before sterilization. Polyethylene terephthalate (PET) sheets were added as the sole carbon source, adapting the protocol described in [22].

2.2 Plastic Sample Conditioning

PET containers were used, which were cut manually under sterility conditions (using scalpel and tweezers) into microplastic sheets of 10.0 x 2.0 cm (pretreated with Tween 80). 5g of plastic samples were available per cell to be exposed to the biodegradation process and bioelectrochemical generation.

2.3 Design, Assembly and Operation of Microbial Fuel Cells (MFCs)

The prototype of the MFCs was built using a 500 mL Pyrex glass bottle with a tight-fitting lid. A 2.5 cm diameter hole was drilled to attach a polyvinyl chloride (PVC) tube of equal diameter and 6.5 cm in length, which functioned as a support for the cathode, while the anode was located at the top of the reactor. Both electrodes were connected by copper wires exposed to the outside to facilitate electrochemical measurements. Previously, the bottle, tube and electrodes were washed, rinsed with distilled water and disinfected with 70%

alcohol. A circular zinc anode of 2.5 cm in diameter and a copper cathode coated with activated carbon of 2.5 × 10 cm were used, previously cleaned and disinfected before assembly. The lid of each cell was equipped with a sterile 0.22 μm PTFE vent filter to ensure passive venting. The reactors operated under constant magnetic stirring at a low speed of 120 rpm.

The experimental evaluation was carried out in MFC with a working volume of 500 mL, using 445 mL of Bushnell–Haas medium, 50 mL of a bacterial consortium and 5 g of plastic as the only carbonate substrate. The consortium was prepared by pre-adjusting each strain to a concentration of 1×10^8 CFU/mL and mixing the standardized cultures in equal ratios (12.5 mL per strain; 1:1:1:1 ratio). The mixture was then incorporated into the culture medium, ensuring a homogeneous concentration of the inoculum in the three experimental units to ensure the reproducibility of the assays.

2.4 Physical-ChemicalMonitoring and Bioelectrochemical Evaluation

During the experimental period, periodic monitoring of the physicochemical and bioelectrochemical parameters of the MFCs was carried out. The physicochemical parameters, including pH and electrical conductivity, were determined by means of an APERA PC60 multiparameter, in order to evaluate the variations of the reaction medium associated with the metabolic activity of the bacterial consortium. Optical density (OD600) was measured with a GENESYS 150 UV-Vis spectrophotometer (Thermo Scientific, Waltham, MA, USA).™. The bioelectrochemical parameters were recorded using a Vernier voltmeter (model DVP 2216R2) and a Vernier current sensor (model HCS-BTA) connected to an external resistance of 100 Ω, obtaining the values of voltage (V) and current (I). Additionally, a Fourier transform infrared spectroscopy (FTIR) analysis of the initial and post-treatment PET with the microbial consortium was performed, using a Thermo Scientific Nicolet iS50 FTIR spectrophotometer (Thermo Fisher Scientific, Madison, WI, USA) in the range of 4000 to 400 cm^{-1}, with a resolution of 4 cm^{-1} and 240 scans per sample. This analysis allowed the identification of the structural changes of the polymer induced by microbial activity. Finally, the pH, conductivity, optical density, voltage and current data were analyzed using descriptive statistics (mean and standard deviation) to characterize the behavior of the system during the experimental process. The trial was performed in triplicate (n=3) to ensure the reproducibility of the results.

III RESULTS AND DISCUSSION

Fig. 1 shows a stable voltage production during the 98 days of operation, with values close to 1.0 V and moderate fluctuations throughout the experiment. These variations are typical of the metabolic dynamics of the biofilm and changes in substrate availability, without showing a sustained loss of the electrochemical performance of the system. This behavior is consistent with a process of microbial colonization of the anode and degradation of the substrate, where hydrolytic bacteria (*Bacillus cereus* and *Actinomyces sp*.) secrete enzymes to depolymerize the PET, releasing monomers that are metabolized by electrogenic bacteria (*P. stutzeri and S. maltophilia).*

Since the inoculum is 10% (50 mL in a total volume of 495 mL), the curve probably starts with a prolonged latency period or lag phase. During this initial phase, the bacteria in the consortium (*Bacillus cereus* and *Actinomyces sp*.) secrete hydrolytic enzymes (such as esterases and cutinases) to depolymerize the plastic, transforming it into bioavailable monomers (short-chain fatty acids) [23,24]. This process is slow due to the recalcitrant nature of the polymer, so the voltage remains close to zero or at very low values for the first few hours or days [25,26].

Once this barrier has been overcome, a pronounced rise in voltage is observed. This is where the synergy of the consortium comes into play: the released monomers are rapidly metabolized by *P. stutzeri* and *S. maltophilia*, exoelectrogenic bacteria par excellence. *P. stutzeri* produces endogenous redox compounds (such as pyoverdine or pyocyanin) that act as natural electronic mediators, facilitating the transfer of electrons from the cell membrane to the anode [27].

This phenomenon explains the sustained increase in voltage, reflecting a mature and electrochemically active biofilm. Since the inoculum is 10% (a moderate concentration), the maximum voltage peak reached stable values between 0.98 – 1.0 V, but without supersaturating the system, which prevents premature depletion of the substrate.

Subsequently, the curve enters a plateau or stationary phase, where the oxidation rate of the metabolites (from the plastic) equals the electrochemical demand. The stability of this plateau is indicative of the robustness of the consortium. However, being a solid substrate (5 g), mass transport is limiting. Over time, the voltage is likely to slowly decay, signaling depletion of the plastic's easily hydrolyzable fraction or accumulation of toxic waste products that inhibit *Actinomyces*. This shows that MFC manages to generate electricity in a sustained way thanks to the metabolic cooperation between hydrolyzers (*Bacillus/Actinomyces*) and electrochemicals(*Pseudomonas/Stenotrophomonas*),suggestig the use of this consortium for the bioremediation of plastic waste.

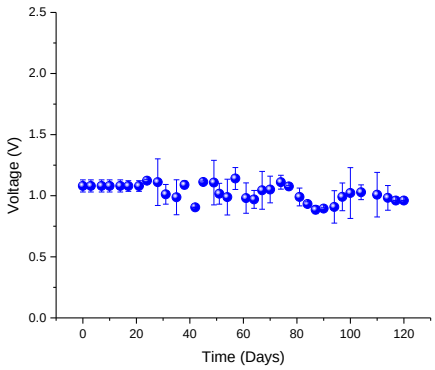


Fig. 1. Voltage values of the microbial fuel cell during the evaluation period.

Fig. 2 shows the electrical current production of the MFC with 5 g of plastic over time. Four phases can be distinguished: 1) Latency phase (days 0-15), with an extremely low and constant current (~0.005 mA), corresponding to the period of adaptation and colonization of the consortium on the anode and the plastic [6,28]. 2) Growth phase (days 15-~30), where the current increases to ~0.03 mA, indicating the onset of polymer chain breakage and electron release [6]. 3) Maximum production phase (~30-45 days), with peaks of 0.05 to 0.056 mA, which show an active degradation of the plastic, sustained by the enzymatic arsenal of *Actinomyces* sp. and *S. maltophilia* [29,30]. The observed fluctuations (decreases to ~0.012 mA) are attributable to metabolic changes or temporary depletion of intermediates [6,31]. Finally, 4) Stabilization phase (days 45-60), where the current stabilizes between 0.035 and 0.04 mA, indicating the maturation of an electroactive biofilm [32]. This stability, together with the structural changes evidenced by FTIR, suggests that the system achieves sustained biodegradation and constant long-term energy recovery.

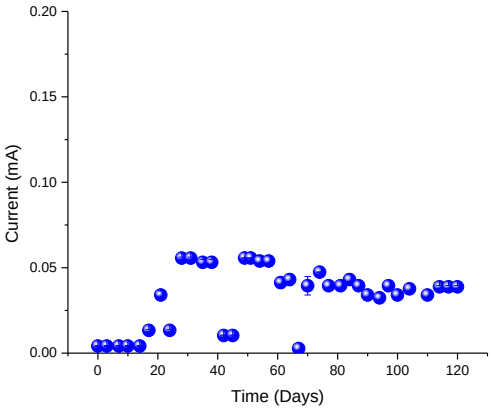


Fig. 2. Current values of the microbial fuel cell during the evaluation period.

Fig. 3 shows the evolution of the suspended biomass (OD_{600}), pH and electrical conductivity of MFC-1 during the first 98 days of operation. OD_{600} increased from 0.017 ± 0.004 at baseline to 0.246 ± 0.023 on day 21, then decreased to 0.142 ± 0.112 on day 49, and finally reached 0.241 ± 0.148 at day 98 (Fig. 3A). This behavior is consistent with an initial phase of adaptation of the bacterial consortium to the system and coincides temporally with the period in which various studies describe the establishment of electroactive biofilms on the anode. The transition of a fraction of the microbial community from the planktonic phase to a sessile state favors the formation of the biofilm, considered the main functional component of microbial fuel cells due to its participation in extracellular electron transfer [33,34]. The use of carbon-coated electrodes may also contribute to this process, as their high electrical conductivity, chemical stability, and biocompatibility provide a suitable surface for microbial adhesion and electron exchange [33,35].

The pH showed a gradual increase from 7.00 ± 0.14 to 9.07 ± 0.45, followed by a slight decrease to 8.37 ± 0.43 at the end of the evaluation period (Fig. 3B).

Similarly, electrical conductivity increased progressively from 2.13 ± 0.22 to 8.89 ± 0.40 mS/cm, with a slight decrease to 7.80 ± 0.87 mS/cm in the last measurement (Fig. 3C). Both parameters reflect physicochemical modifications compatible with the metabolic activity of the bacterial consortium. A slightly alkaline pH favors the enzymatic activity and stability of the electroactive communities, while the increase in conductivity facilitates the transport of protons and other ions between the electrodes, decreasing internal resistance and providing favorable conditions for charge transfer [33,35]. Taken together, the evolution of these parameters is consistent with the progressive establishment of a functional bioelectrochemical system, as described for microbial fuel cells operated with electroactive communities [34,35].

The observed changes in suspended biomass and physicochemical parameters describe the operational evolution of the MFC during the first 98 days of operation. Although these results show the establishment of the bacterial consortium and conditions compatible with the development of bioelectrochemical activity, the comprehensive interpretation of the process requires considering together the structural characterization of PET by FTIR and the electrochemical response of the system. The integration of these evidences will allow a more accurate assessment of the interaction between the bacterial consortium, the polymer and the functioning of the Pourhosseini microbial fuel cell [6].

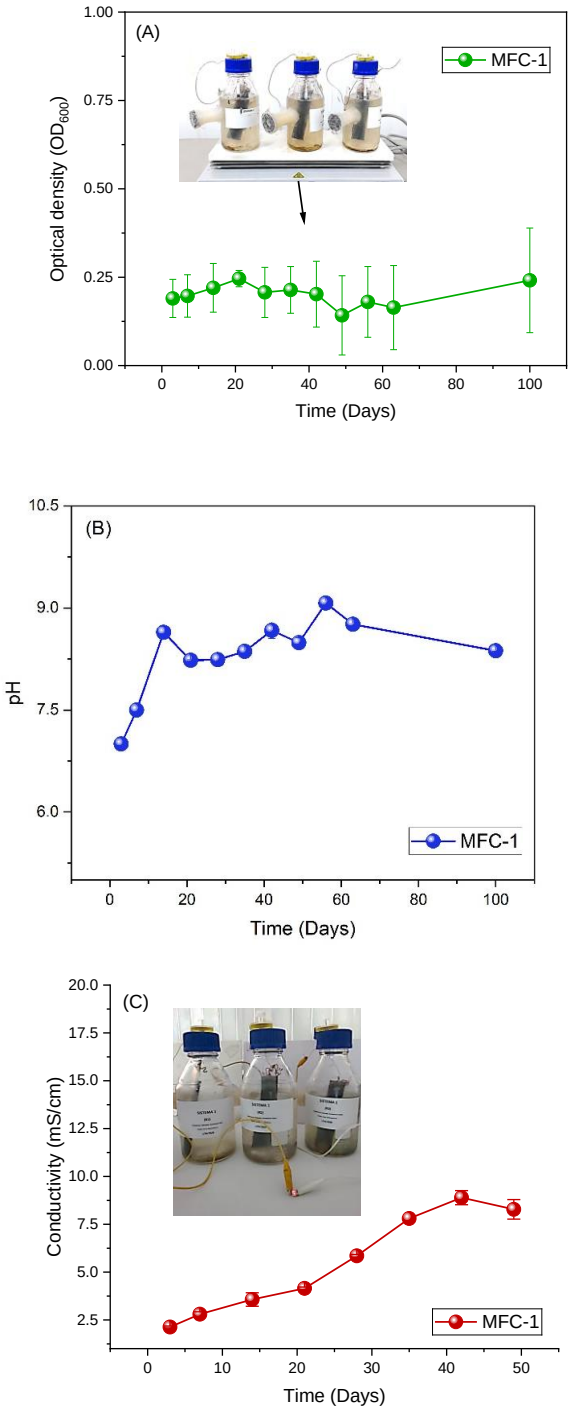


Fig. 3. Temporal evolution of suspended biomass, pH and electrical conductivity in MFC-1 during the evaluation period. (A) Suspended biomass determined by optical density at 600 nm (OD600). (B) pH variation. (C) Electrical conductivity. Data represent the mean ± standard deviation (n = 3).

Fig. 4 shows the FTIR spectrum of the initial PET plastic (blue) and the one treated with the consortium (red). In the treated spectrum, a relevant change in the absorption band is observed at 3411 cm^{-1}, corresponding to the stretching of hydroxyl (-OH) groups, which suggests a greater exposure of these groups and suggests the incorporation of oxygen in the polymeric matrix by enzymatic oxidation. Likewise, the decrease in transmittance in the regions of 2900–2800 cm^{-1} (C–H), 1700–1600 cm^{-1} (C=O), and 1200–900 cm^{-1} (C–O–C) indicates changes in functional composition and glycosidic bonds.

Specifically, the increase in band intensity in the range of ~1715 cm^{-1} (assigned to esters or carboxylic acids) is consistent with polymer biooxidation [36]. While the initial PET shows definite peaks of the intact structure, the treated sample shows a reduction in the intensity of these bands, suggesting cleavage of primary chemical bonds [37]. Taken together, these spectral changes are indicative that the microbial consortium is chemically modifying the plastic, functionalizing its surface, and causing alterations in the polymer structure.

In all three MFCs, these spectral alterations suggest that the consortium is metabolizing the plastic substrate for use as a carbon source and electron donor. Strains of *Bacillus cereus* have been reported to possess the ability to degrade polymers by hydrolytic enzymes [30,38]. On the other hand, *Stenotrophomonas maltophilia* and members of the Actinomyketata (*Actinomyces* sp.) act efficiently in the fragmentation of complex hydrocarbons and recalcitrant polymers, facilitating the transfer of electrons to the anode of the cell [39].

Results of the FTIR analysis are indicative of possible structural changes in the PET that could be associated with biodegradation processes and is could be related to the generation of bioelectricity in the MFC, suggesting the potential efficiency of the system for simultaneous bioremediation and energy recovery [6]. However, it is important to note that the FTIR provides information on changes in the chemical composition of the surface, but does not allow the evaluation of morphological damage or loss of structural integrity of the polymer. Therefore, the absence of complementary analyses, such as scanning electron microscopy (SEM), which would have confirmed surface erosion, cracking or reduction in PET particle size, is recognized as a limitation of the present study. It is recommended that future studies incorporate these techniques to comprehensively validate the physical degradation of plastic.

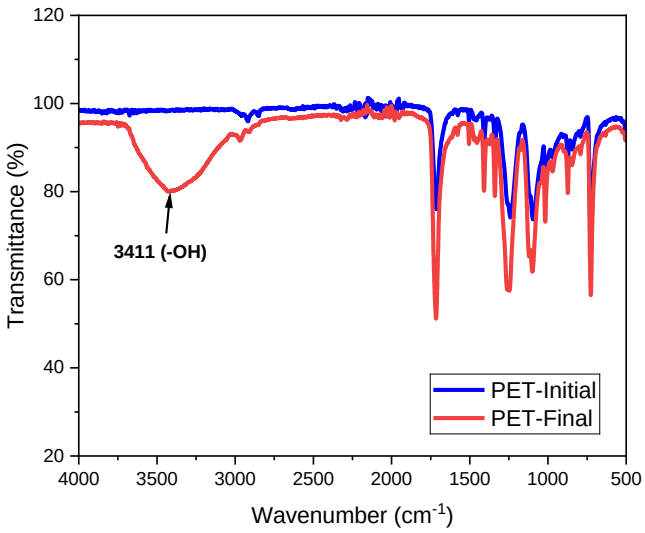


Fig. 4. FTIR spectra of the initial PET and PET treated with the microbial consortium.

From a circular economy perspective, the results of this study suggest that PET waste can be transformed from an environmental liability into a value-added energy resource. The simultaneous biodegradation of PET and bioelectricity generation in a single-chamber MFC represents a promising platform for green innovation and sustainable entrepreneurship. Unlike conventional recycling, which requires high energy inputs and generates toxic by-products, this bioelectrochemical approach offers a low-cost, environmentally friendly alternative that aligns with the principles of the circular economy. The scalability of this system could enable the development of decentralized waste-to-energy solutions, particularly in regions with inadequate plastic waste management infrastructure. Furthermore, the integration of this technology with existing recycling streams could create new business models based on the valorization of plastic waste, contributing to the transition towards a more sustainable and resource-efficient economy.

IV CONCLUSIONS

The bacterial consortium made up of *Pseudomonas stutzeri*, *Bacillus cereus*, *Stenotrophomonas maltophilia and Actinomyces sp*. demonstrated synergistic capacity with potential for the biodegradation of PET and the simultaneous generation of bioelectricity in a single-chamber microbial fuel cell, using plastic as the only carbon source. The system reached a maximum voltage of approximately 1.0 V and a peak current of 0.056 mA, exceeding the values reported in previous research using activated sludge (222 mV) and suggesting that the selection of a consortium with hydrolytic and electrogenic bifunctional capabilities significantly improves the electrical performance of the MFC. The FTIR analysis evidenced the biooxidation of PET by increasing hydroxyl (-OH) and carbonyl (C=O) groups, as well as the decrease in the intensity of C-H and C-O-C bonds, structural changes that suggest the cleavage of polymer chains attributable to the enzymatic action of *Bacillus cereus and Actinomyces sp*. Likewise, the evolution of pH (from 7.0 to 9.07), electrical conductivity (from 2.13 to 8.89 mS/cm) and suspended biomass (OD_{600}) was consistent with the progressive establishment of an active microbial community and the formation of electroactive biofilms, conditions that favored extracellular electron transfer and the operational stability of the system during the 98 days of monitoring. These findings address the scientific gap identified in the literature by demonstrating that the optimization of a specific bacterial consortium and the control of operating conditions allow the effective integration of PET biodegradation with bioelectricity generation, laying the foundations for the development of sustainable technologies aligned with the principles of the circular economy.

Therefore, it is recommended that future studies complement these findings with morphological characterization techniques such as scanning electron microscopy (SEM) and atomic force microscopy (AFM), which allow the evaluation of erosion, cracking and loss of structural integrity of PET. It is also suggested to evaluate the effect of different substrate concentrations, explore the scalability of the system and perform an analysis of the anodic biofilm microbiome to identify the genetic mechanisms responsible for the degradation of PET, as well as incorporate advanced electrochemical techniques to characterize the kinetics of interfacial processes.

AGRADECIMIENTO/RECONOCIMIENTO

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REFERENCES

[1] PlasticsEurope, Plastics—The Facts 2020: An Analysis of European Plastics Production, Demand and Waste Data. Brussels, Belgium: PlasticsEurope, 2020. [Online]. Available: https://plasticseurope.org/knowledge-hub/plastics-the-facts-2020/. [Accessed: Jul. 5, 2022].

[2] J. Yuan, J. Ma, Y. Sun, T. Zhou, Y. Zhao, and F. Yu, "Microbial degradation and other environmental aspects of microplastics/plastics," Sci. Total Environ., vol. 715, Art. no. 136968, 2020.

[3] C. M. Rochman et al., "Classify plastic waste as hazardous," Nature, vol. 494, no. 7436, pp. 169–171, Feb. 2013, doi: 10.1038/494169a.

[4] N. Mohanan, Z. Montazer, P. K. Sharma, and D. B. Levin, "Microbial and enzymatic degradation of synthetic plastics," Front. Microbiol., vol. 11, Art. no. 580709, 2020, doi: 10.3389/fmicb.2020.580709.

[5] S. Kim et al., "Bioelectrochemical enhancement of polybutylene succinate degradation via electricity-supplemented engineered Shewanella oneidensis MR-1," Bioelectrochemistry, vol. 168, Art. no. 109132, Mar. 2026, doi: 10.1016/j.bioelechem.2025.109132.

[6] P. S. Pourhosseini, M. Giyahchi, S. Danaee, and H. Moghimi, "Using dual chamber microbial fuel cells for coupled microplastic biodegradation and bioelectricity production: Assessing the effect of substrate," *Microb. Cell Fact.*, vol. 25, Art. no. 52, 2026, doi: 10.1186/s12934-026-02925-y.

[7] S. Joo et al., "Structural insight into molecular mechanism of poly(ethylene terephthalate) degradation," Nat. Commun., vol. 9, no. 1, Art. no. 382, 2018.
[8] A. Hadji-Thomas, S. Wang, Y. Gariepy, and V. Raghavan, "The exploitation of single-chambered microbial fuel cells for PET removal in water," *Microorganisms*, vol. 13, no. 11, Art. no. 2500, Oct. 2025, doi: 10.3390/microorganisms13112500
[9] S. Kalathil, M. Miller, and E. Reisner, "Microbial fermentation of polyethylene terephthalate (PET) plastic waste for the production of chemicals and electricity," *ChemRxiv*, 2021, doi: 10.26434/chemrxiv-2021-2cctp.
[10] A. Tirkey and L. S. B. Upadhyay, "Introducing the LDPE degrading microbes of sedimentary systems: From dumpsite to laboratory," *Environ. Sci.: Adv.*, vol. 4, no. 6, pp. 952–963, 2025, doi: 10.1039/D5VA00058K.
[11] K. Kaur, S. Sharma, N. Shree, and R. Mehrotra, "Recent advancements and mechanism of plastics biodegradation promoted by bacteria: A key for sustainable remediation for plastic wastes," *Biosci. Biotechnol. Res. Asia*, vol. 20, no. 1, pp. 1–12, Mar. 2023.
[12] T. C. Dakal *et al.*, "New horizons in microbial fuel cell technology: Applications, challenges, and prospects," *Biotechnol. Biofuels Bioprod.*, vol. 18, Art. no. 79, 2025, doi: 10.1186/s13068-025-02649-y.
[13] B. E. Logan, R. Rossi, A. Ragab, and P. E. Saikaly, "Electroactive microorganisms in bioelectrochemical systems," *Nat. Rev. Microbiol.*, vol. 17, no. 5, pp. 307–319, May 2019, doi: 10.1038/s41579-019-0173-x.
[14] S. S. S. Garimella, S. V. Rachakonda, S. S. Pratapa, G. D. Mannem, and G. Mahidhara, "From cells to power cells: Harnessing bacterial electron transport for microbial fuel cells (MFCs)," *Ann. Microbiol.*, vol. 74, no. 1, Art. no. 19, Jun. 2024, doi: 10.1186/s13213-024-01761-y.
[15] P. Li *et al.*, "Complete genome sequence of *Pseudomonas stutzeri* S116 owning bifunctional catalysis provides insights into affecting performance of microbial fuel cells," *BMC Microbiol.*, vol. 22, Art. no. 137, 2022, doi: 10.1186/s12866-022-02552-8.
[16] C. K. Varnava, P. Persianis, I. Ieropoulos, and A. Tsipa, "Electricity generation and real oily wastewater treatment by *Pseudomonas citronellolis* 620C in a microbial fuel cell: Pyocyanin production as electron shuttle," *Bioprocess Biosyst. Eng.*, vol. 47, no. 6, pp. 903–917, Jun. 2024, doi: 10.1007/s00449-024-03016-1.
[17] S. Rojas-Flores, M. De La Cruz-Noriega, N. M. Otiniano, L. Cabanillas-Chirinos, and L. M. Angelats-Silva, "Electricity generation and plastic waste reduction using the fungus *Paecilomyces* as a biodegrader in microbial fuel cells," *Sustainability*, vol. 16, no. 24, Art. no. 11137, Dec. 2024, doi: 10.3390/su162411137.
[18] X. Qi, X. Gao, X. Wang, and P. Xu, "Harnessing *Pseudomonas putida* in bioelectrochemical systems," *Trends Biotechnol.*, vol. 42, no. 7, pp. 877–894, Jul. 2024, doi: 10.1016/j.tibtech.2023.12.002.
[19] W. Rojas-Villacorta *et al.*, "Potential use of pepper waste and microalgae *Spirulina* sp. for bioelectricity generation," *Energy Rep.*, vol. 9, pp. 253–261, 2023, doi: 10.1016/j.egyr.2023.09.007.
[20] S. Rojas Flores et al., "Agricultural wastes for electricity generation using microbial fuel cells," Open Biotechnol. J., vol. 14, pp. 52–58, 2020, doi: 10.2174/1874070702014010052.
[21] S. Bisht, P. Pandey, B. Bhargava, S. Sharma, V. Kumar, and K. D. Sharma, "Bioremediation of polyaromatic hydrocarbons (PAHs) using rhizosphere technology," Braz. J. Microbiol., vol. 46, no. 1, pp. 7–21, Mar. 2015, doi: 10.1590/S1517-838246120131354
[22] R. Usha, T. Sangeetha, and M. Palaniswamy, "Screening of polyethylene degrading microorganisms from garbage soil," *Libyan Agric. Res. Cent. J. Int.*, vol. 2, no. 4, pp. 200–204, 2011.
[23] A. K. Prajapati et al., "Harnessing waste for energy: Feedstock, technological advancement, sustainability, life cycle evaluation, and future perspectives," Fuel, vol. 410, Art. no. 137965, 2026, doi: 10.1016/j.fuel.2025.137965.
[24] K. Rabaey, N. Boon, M. Höfte, and W. Verstraete, "Biofuel cells select for microbial consortia that self-mediate electron transfer," *Appl. Environ. Microbiol.*, vol. 71, no. 4, pp. 2019–2026, Apr. 2005.
[25] H. Zhao *et al.*, "Off-grid circular upcycling of polyimide plastic waste into hydrogen, monomers, and protein via solar-electro-bioprocessing," *Appl. Catal. B: Environ. Energy*, vol. 392, Art. no. 126710, 2026, doi: 10.1016/j.apcatb.2026.126710.
[26] B. E. Logan *et al.*, "Microbial fuel cells: Methodology and technology," *Environ. Sci. Technol.*, vol. 40, no. 17, pp. 5181–5192, Sep. 2006, doi: 10.1021/es0605016.
[27] M. Liao *et al.*, "Construction of conductive modified polyethylene terephthalate-supported cathode in microbial fuel cell for nitrate removal," *Bioresour. Technol.*, vol. 445, Art. no. 134050, 2026, doi: 10.1016/j.biortech.2026.134050.
[28] R. Lyons et al., "The diversity of plastisphere bacterial and fungal communities differs between biodegradable polymer types in soil," Microb. Ecol., vol. 89, Art. no. 56, 2026, doi: 10.1007/s00248-025-02677-z.
[29] K. Naseer *et al.*, "Isolation of aerobic denitrifying bacteria *Stutzerimonas stutzeri* and its application in coking wastewater treatment," *Sci. Rep.*, vol. 16, Art. no. 8717, 2026, doi: 10.1038/s41598-026-43338-6.
[30] S. Y. Yang et al., "Zero valent iron-assisted methane production from anaerobic digestion of microalgae Tetradesmus obliquus," Appl. Biol. Chem., vol. 69, Art. no. 20, 2026, doi: 10.1186/s13765-026-01089-y.
[31] E. A. E. Sakr, M. S. Hassan, M. El-Sedik, M. M. Rekaby, and G. M. El-Sayed, "Residual-hybrid dynamic response-surface/machine-learning framework for predicting malachite green removal and biodegradation by a bacterial consortium," *J. Biol. Eng.*, vol. 20, Art. no. 64, 2026, doi: 10.1186/s13036-026-00663-8.
[32] A. Chaudhary *et al.*, "Performance assessment of a PUF integrated anaerobic bioreactor for textile effluent decolourization along with lab to pilot scale transition and toxicity assessment," *Discover Sustainability*, vol. 7, Art. no. 198, 2026, doi: 10.1007/s43621-025-02549-z.
[33] A. Nawaz, N. V. Singh, S. Samuchiwal, *et al.*, "A state of the art review on electron transfer mechanisms, characteristics, applications and recent advancements in microbial fuel cells technology," *Green Chem. Lett. Rev.*, vol. 13, no. 4, pp. 365–381, 2020, doi: 10.1080/17518253.2020.1854871.
[34] R. Chakma *et al.*, "Recent applications, challenges, and future prospects of microbial fuel cells: A review," *Global Challenges*, vol. 9, no. 5, Art. no. 2500004, 2025, doi: 10.1002/gch2.202500004.
[35] A. M. Nasrabadi, "A detailed survey of microbial fuel cells: Classifications, computational modeling, recent innovations, and emerging applications," Renew. Sustain. Energy Rev., vol. 226, Art. no. 116241, 2026, doi: 10.1016/j.rser.2026.116241.
[36] P. Sahu *et al.*, "Polyurethane biodegradation by *Fusarium parceramosum* with insights into kinetic modelling and metabolic pathway," *Bioresour. Bioprocess.*, vol. 13, Art. no. 82, 2026, doi: 10.1186/s40643-026-01079-4.
[37] R. M. Kathrotiya et al., "Statistical optimization and metabolite analysis of Acid Yellow 137 dye biodegradation by bacterial consortium," Sci. Rep., vol. 16, Art. no. 20108, 2026, doi: 10.1038/s41598-026-49978-y.
[38] P. Kulshrestha *et al.*, "Multi copper oxidase from *Micrococcus* sp. IITD107, a promising candidate for polypropylene biodegradation," *npj Mater. Degrad.*, vol. 10, Art. no. 8, 2026, doi: 10.1038/s41529-025-00720-5.
[39] J. Marciniak et al., "Nocardia fodinahabitans sp. nov. isolated from underground hard coal mine waters shows biotechnological potential for degradation of aromatic hydrocarbons," Mol. Genet. Genomics, vol. 301, Art. no. 133, 2026, doi: 10.1007/s00438-026-02456-6.