Journal of Environmental Chemical Engineering 14 (2026) 123049

Contents lists available at ScienceDirect

Journal of Environmental Chemical Engineering

journal homepage: www.elsevier.com/locate/jece

Biosynthesis of polyhydroxyalkanoate bioplastics using waste-derived volatile fatty acids

Milica Mihajlovic [a], Kaveh Shahbaz [b], Céline Vaneeckhaute [c,d], Saeid Baroutian [a,*]

[a] Department of Chemical and Materials Engineering, The University of Auckland, Auckland 1010, New Zealand
[b] College of Science and Engineering, James Cook University, Townsville, Queensland 4811, Australia
[c] BioEngine, Research Team on Green Process Engineering and Biorefineries, Chemical Engineering Department, Université Laval, 1065 Avenue de La Médecine, Québec, QC G1V 0A6, Canada
[d] CentrEau/c-Eau Centre de Recherche sur L'eau, Université Laval, 1065 Avenue de La Médecine, Québec, QC G1V 0A6, Canada

ARTICLE INFO

Keywords:
Polyhydroxyalkanoates
Volatile Fatty Acids
Bioplastics
Circular Bioeconomy

ABSTRACT

The widespread environmental and health issues associated with petroleum-derived plastics have prompted the search for sustainable alternatives, positioning polyhydroxyalkanoates (PHAs) as a particularly promising class of biocompatible and fully biodegradable polyesters. Despite their favourable properties, high production costs, mainly stemming from the expensive conventional carbon substrates, currently impede the industrial-scale up of PHAs. This review highlights that waste-derived volatile fatty acids (VFAs) serve as strategically viable precursors for PHA biosynthesis, not only for cost reduction but for their unique metabolic advantages. Utilising VFAs shortens the microbial metabolic pathway, leading to high substrate-to-PHA conversion efficiencies, with yields reaching up to 91% dry cell weight in optimised systems. Crucially, the key strength of this pathway is that control of VFA composition (e.g., the propionate-to-acetate ratio) enables tailoring of the polymer monomer profile and, consequently, its mechanical properties for different applications. However, this review highlights the central bottleneck to industrialisation as the trade-off between this metabolic efficiency and the inherent microbial toxicity of concentrated VFAs. A comparative analysis against conventional substrates highlights the significant environmental and economic advantages of the VFA pathway, including lower cumulative energy demand and gate-free incentives associated with traditional carbon sources.

1. Introduction

The widespread use of petroleum-derived plastics has become a subject of increasing controversy due to their profound impacts on ecosystems and human health. Global plastic production has escalated dramatically, reaching approximately 413.8 million metric tonnes annually in recent years, with projections suggesting continued exponential growth in the coming decades [1,2]. Conventional petroleum-based plastics are highly resistant to degradation, persisting for hundreds of years and fragmenting into microplastics that infiltrate food chains, water systems, and even human tissues, posing significant ecological and health risks [3–5]. The drastic health and environmental consequences of plastic pollution have driven increased research into innovative solutions, particularly biodegradable plastics. Polyhydroxyalkanoates (PHAs) have emerged as a particularly promising alternative. They are classified as microbial polyesters and are accumulated by various PHA-producing microorganisms as intracellular energy storage compounds under nutrient-limited and carbon-excess conditions [6–8]. Unlike many other bioplastics, PHAs have been repeatedly shown to degrade in a wider array of microbial-rich natural environments, including marine and soil ecosystems. In addition to their advantageous biodegradability, they exhibit adjustable properties comparable to those of conventional polymers, making them suitable for various applications in packaging, agriculture, and medicine [9–11]. Depending on the specific type of PHA, they can mimic the strength, stiffness, and elasticity of common plastics, such as polypropylene, making them versatile for packaging and consumer goods. They possess non-toxic and biocompatible properties, making them suitable for medical applications such as sutures, implants, and drug delivery systems as highlighted in Table 1.

However, despite their advantageous biodegradability and functionality, PHAs face significant barriers to commercialisation, primarily

* Corresponding author.
E-mail address: s.baroutian@auckland.ac.nz (S. Baroutian).

https://doi.org/10.1016/j.jece.2026.123049
Received 21 February 2026; Received in revised form 5 May 2026; Accepted 5 May 2026
Available online 8 May 2026
2213-3437/© 2026 The Author(s). Published by Elsevier Ltd. This is an open access article under the CC BY license (http://creativecommons.org/licenses/by/4.0/).

due to high production costs [10]. These costs stem from the expensive substrates required for microbial fermentation and the relatively low yields compared to petrochemical-derived plastics [22]. Typical PHA production costs range from ~5–15 USD/kg of PHA, depending on scale, substrate, and recovery method, compared with conventional petroleum-based plastics, which cost between 2–3 USD/kg of plastic [23]. The primary expenses originate from carbon substrates and downstream processing, including extraction and purification techniques, which can account for over 50% of the total cost [12]. Despite their environmental promise, these economic disparities have limited PHAs' competitiveness in the global plastics market. Consequently, current research efforts are focused on reducing production costs by identifying and utilising low-cost carbon sources.

One strategy currently gaining significant attention in sustainable biopolymer research is the valorisation of volatile fatty acids (VFAs) as substrates for PHA production. Classified as a group of short-chain organic acids, including acetic, propionic, butyric, and valeric acids, VFAs are not only crucial intermediates in waste management processes such as anaerobic digestion and fermentation but also versatile building blocks for the biochemical and biofuel industries [24]. A key advantage of VFAs lies in their widespread availability, as they can be sustainably produced through treatment of a wide range of organic wastes, including municipal sludge, food waste, agricultural residues, and agro-industrial by-products [24,25]. Compared to sugar-based substrates that require saccharification, VFAs can offer a more direct carbon source for PHA production. However, waste-derived VFA liquors often contain impurities such as ammonia, sulphides, metals, surfactants, and phenolics, meaning that polishing or other pre-treatment steps may still be necessary to remove inhibitory compounds. Notably, the distribution of VFA chain lengths influences both microbial uptake efficiency and the type of PHAs produced (summarised in Table 2), affecting polymer composition and final material properties, resulting in polymers with

Table 1
PHA variations available in the literature and the carbon sources used for production.

PHA type		Monomers	Material Properties*	Applications	Carbon sources	Ref.
Poly(3-hydroxybutyrate)	PHB	3-hydroxybutyrate	Tensile strength = ~20–40 MPa Young's modulus = 1.2–2.2 GPa Elongation at break = ~1–8% Melting Temp. = ~160–180 °C	Packaging, drug delivery, and tissue engineering	Glucose, sucrose, VFAs (acetic acid)	[12, 13]
Poly(3-hydroxyvalerate)	PHV **	3-hydroxyvalerate	Tensile strength = ~31 MPa Young's modulus = 1.5–6 GPa Elongation at break = > 14% Melting temp. = ~ 119 °C	Films and coatings	Propionic acid, valeric acid, acetic acid	[14]
Poly(3-hydroxybutyrate-co-3-hydroxyvalerate)***	PHBV	3-Hydroxybutanoic acid & 3-Hydroxyvaleranoic acid	Tensile strength = ~10–30 MPa Young's modulus = 1–6 GPa Elongation at break = >50% Melting Temp. = ~160–180 °C	Packaging, agricultural films, and drug delivery	Glucose, valeric acid, acetic acid	[15]
Poly-3-hydroxypropionate	PHP	3-hydroxypropionate	Tensile strength = ~27 MPa Young's modulus = 0.3 GPa Elongation at break = > 19–342% Melting temp. = ~ 77 °C	Packaging and agricultural films	Propionate, Glycerol, Propanol	[16, 17]
Poly(3-hydroxyhexanoate)	PHHx	Variable Hydroxyalkanoic acids	Tensile strength = ~20–22 MPa Young's modulus = ~ 0.9–1.2 GPa Elongation at break = > 100% Melting temp. = ~ 100–150 °C	Adhesives, coatings and medical materials	Sugars, fatty acids, alkanes, plant oils, food/industrial wastes	[18, 19]
Poly(3-hydroxyoctanoate)	PHO	3-hydroxyoctanoate	Tensile strength = ~3–7 MPa Young's modulus = 1.8–17 MPa Elongation at break = >250% Melting temp. = 54–62 °C	Drug delivery, tissue engineering, and flexible films	Long-chain fatty acids, alkanes	[20, 21]

* Literature on specific PHAs and their properties is very limited; hence, the material properties stated in this table are an estimate and may vary depending on future research and publications.

** PHV is rarely found as a homopolymer and is mostly synthesised as PHBV from currently available literature. If pure, it is expected to have properties similar to those of PHB.

*** Semicrystalline copolymer with properties strongly dependent on 3HV fraction; increasing 3HV generally lowers crystallinity and melting temperature, decreases stiffness/brittleness, and improves ductility/toughness.

Table 2
Types of VFAs used for PHA production.

VFA	Chemical Structure	Associated PHAs	Typical Polymer Type	Ref.
Acetic acid	CH_3COOH	PHB	Homopolymer	[28]
Valeric acid	$CH_3(CH_2)_3COOH$	PHV, PHBV	Copolymer	[28]
Butyric acid	$CH_3CH_2CH_2COOH$	PHB	Homopolymer	[28]
Propionic acid	CH_3CH_2COOH	PHV, PHBV	Copolymer	[28]

adjustable properties and applications [24,26]. For instance, acetic and butyric acids are often associated with PHB production, while longer-chain VFAs, such as valeric acid, promote copolymer formation, thereby improving material flexibility. This versatility highlights VFAs not only as economical but also as adjustable feedstocks for tailoring PHAs with desired properties [27].

Current trends in literature highlight the suitability of VFAs as PHA substrates, their economic advantages, and environmental impacts, demonstrating significant promise in lowering PHA production costs and enhancing commercial potential [6,13,29]. While several studies highlight a sustainable and realistic approach to commercial PHA production, there is a lack of discussion regarding scale-up and process integration challenges associated with waste-derived VFAs as PHA substrates (seen in Table 3). When using waste-derived materials, several challenges arise related to compositional variability, process stability, and downstream processing [6]. The heterogeneity of waste streams, stemming from fluctuations in organic content, pH, and inhibitory compounds such as ammonia or heavy metals, can significantly affect both the VFA yield and composition, leading to unpredictable PHA accumulation and polymer quality, which is often underlined in many review articles covering PHA synthesis from VFAs [6,30]. Recent reviews have extensively explored the technical feasibility of using VFAs for PHA synthesis and the impact of VFA chain length on polymer composition. However, a critical gap remains in the literature regarding the integrated challenges of process scale-up and the lack of robust comparisons between waste-derived VFAs and conventional glucose-based substrates in terms of life-cycle environmental metrics, such as carbon footprint and water usage. This review outlines a sustainable strategy for producing PHAs using VFAs extracted from various waste streams to address these gaps. This approach is crucial for mitigating the high production costs that currently hinder the industrial-scale production of PHAs. Using waste streams for PHA production offers a synergistic solution that simultaneously addresses waste management challenges and reduces the cost of biopolymer synthesis. The valorisation of organic waste into valuable bioproducts, such as PHAs, aligns well with the principles of a circular economy, promoting resource efficiency, reducing reliance on virgin raw materials, and mitigating environmental impacts.

2. Generation of volatile fatty acids from waste streams

The growing interest in VFAs stems from their potential to replace petrochemical-derived feedstocks with sustainable alternatives, aligning with circular economy principles and advancing waste valorisation strategies. Primarily comprising acetic, propionic, butyric, and valeric acids, VFAs are short-chain carboxylic acids (C2–C5) that are primarily produced via anaerobic digestion of organic matter [24,25]. The generation of VFAs from waste streams represents a critical upstream step that directly determines their suitability as carbon substrates for PHA biosynthesis (illustrated in Fig. 1). Specifically, the production pathway, operational conditions, and resulting VFA composition influence key parameters, including monomer distribution, impurity profile, and microbial compatibility, which in turn govern PHA yield, composition, and material properties. Therefore, this section focuses not on VFA production as an independent objective, but on how different generation routes shape the downstream performance of VFA-to-PHA conversion systems [24].

Table 3
Trends in literature demonstrating research available on VFA utilisation for PHA production.

Ref.	Review highlights	Required further work
[13]	– VFAs are identified as a sustainable feedstock for PHA synthesis – Discussion of microbial selection and its impacts on PHA type – Explore the economic and environmental benefits of utilising VFAs	– Discussion of scale-up and process integration challenges – Exploration of VFA composition control and microbial strain optimisation
[6]	– VFA composition and chain length impacts on PHAs. – Discussion of how diverse microbial strains have an impact on PHAs produced from VFAs. – Fermentation optimisation strategies include pH adjustments, organic loading rates, C:N ratios.	– Impurities and variable VFA composition from waste streams inhibit microbial growth and PHA yield. – Discussion of VFA variability and how it may impact the PHA production yields and efficiencies. – Discussion of scale-up issues and economic and market outlooks. – Comparison of utilising conventional carbon sources with waste-derived VFAs.
[29]	– Latest technologies for converting the organic waste into multiple biofuels. – Organic acids, bioplastics, and biocompost production from organic waste. – Techno-economic analysis for organic waste.	– Analysis of the challenges and barriers in integrating different waste streams for optimal bioprocessing. – Discussion on the economic and technical feasibility of scaling up the organic waste valorisation processes to industrial levels.
[30]	– Feasibility of PHA generation from dark fermentation-derived VFAs from organic waste streams. – Review of the characteristics and application of PHAs. – Explores the circular bioeconomy potential and contribution to circular economy goals by converting waste organic streams into high-value bioplastics.	– Control microbial consortia behaviour, selection, and adaptation to mixed VFAs for stable long-term operation. – Economic assessment and feasibility of PHA production from fermentation effluents. – Discussion of composition variability and its effect on the final PHAs produced.
[31]	– Explores the potential of agricultural waste as a low-cost carbon source. – Details different pretreatment methods and hydrolysis routes for converting complex biomass into usable substrates.	– Discussion of the high pretreatment costs and the potential of inhibitors. – Discussion of carbon footprint, water usage, energy inputs, etc., to compare waste-based PHA vs conventional plastics robustly.
[32]	– Conditions of PHA production and microbes required/capable of converting biowaste into PHAs. – Economic assessment of PHA production. – Functionalisation of PHAs and the potential to increase – their application variability.	– Discussion of how variability in biowaste composition may impact microbial growth and polymer yields.

2.1. VFA production pathways

Conventionally synthesised from petrochemicals via chemical oxidation or other chemical processes, production of VFAs (for instance, acetic acid) is often overwhelmingly dependent on fossil resources [33]. The vast majority of the global acetic acid output (~85%) is produced by synthetic petrochemical routes (primarily methanol carbonylation) [34–36]. In contrast, bio-based routes currently supply only a minor fraction of the market, accounting for ~10–15% or less of total production [34]. The primary concern lies in the environmental strain associated with fossil-derived VFA production. Conventional

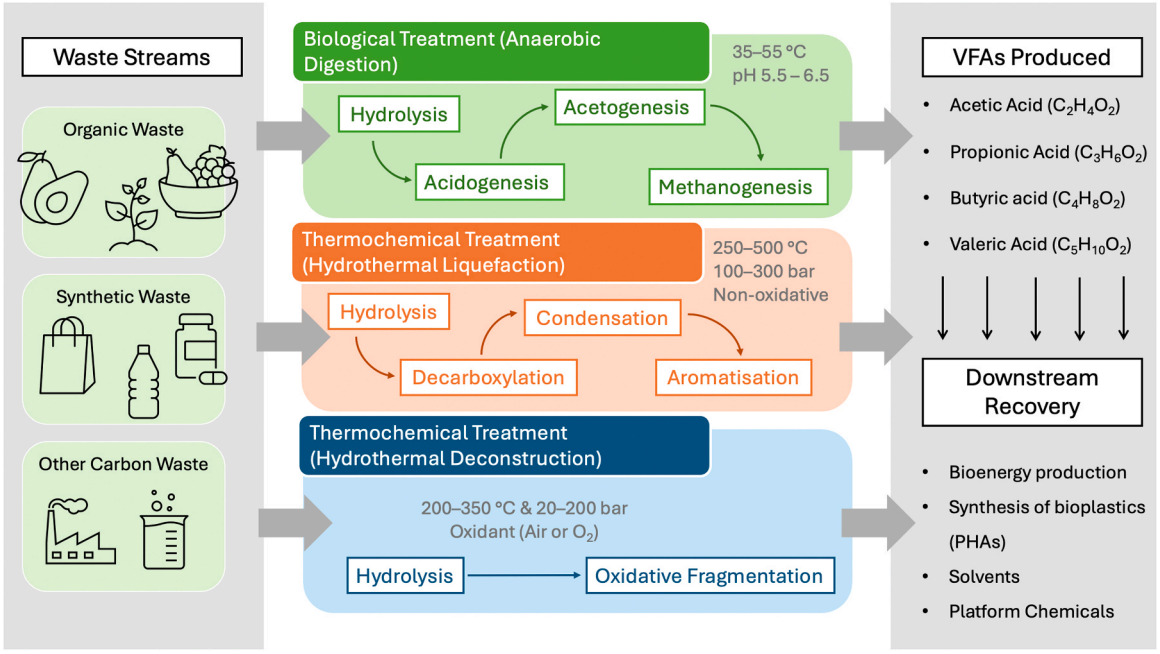


Fig. 1. VFA production methods from various waste streams via biological and thermochemical processing methods.

fossil-based processes are known to produce high yields of VFAs, with relatively fast production rates. For instance, acetic acid is primarily produced through the carbonylation of methanol, while propionic and butyric acids are synthesised via hydrocarboxylation and oxo processes, respectively [24]. These routes offer high product selectivity and purity, which underpins their widespread industrial adoption in applications ranging from plastics and textiles to food and pharmaceuticals. However, they require elevated temperatures and pressures, rely on non-renewable carbon sources, generate significant upstream and process-related greenhouse gas emissions, and remain tied to volatile fossil fuel markets, contributing to both economic and environmental challenges [24]. In contrast, various biological and thermochemical treatment methods of organic waste or biomass residues have been proven effective as research on VFA production has developed over the years, each offering distinct advantages depending on the feedstock composition and the desired VFA profile. They offer the potential to reduce life-cycle emissions and improve circularity.

Overall, the significance of VFA production pathways in the context of PHA biosynthesis lies not only in total acid yield, but also in the resulting acid spectrum, impurity burden, and consistency of the recovered liquor. Biological fermentation routes are generally more favourable when the objective is to produce VFA mixtures readily usable for microbial PHA synthesis, as they can achieve relatively high VFA concentrations under milder conditions. In contrast, thermochemical routes such as hydrothermal liquefaction (HTL) and hydrothermal deconstruction (HTD) may be advantageous for difficult or heterogeneous wastes, but they often generate lower acid concentrations, broader impurity profiles, or oxygenated by-products that increase downstream purification requirements. Therefore, the choice of VFA production route directly affects subsequent microbial uptake, attainable PHA composition, process stability, and overall industrial feasibility.

2.1.1. *Biological treatment methods*

Anaerobic digestion (AD) of organic matter remains the most widely studied biological route, leveraging microbial cultures to convert organic waste streams into VFAs under controlled conditions [33]. This process involves a series of microbial-driven biochemical steps, including hydrolysis, acidogenesis, acetogenesis, and methanogenesis [37]. During hydrolysis, complex organic matter (proteins, carbohydrates, and lipids) in waste is broken down into soluble compounds by extracellular enzymes, as demonstrated in Eqs. 1, 2 and 3 [38].

$$RCH(NH_2)COOH \rightarrow \quad NH_2CH(R)COOH \tag{1}$$

$$(C_6H_{10}O_5)_n \quad \rightarrow \quad C_6H_{12}O_6 \tag{2}$$

$$(RCOO)_3C_3H_5 \quad \rightarrow \quad C_3H_{12}O_3 + RCOOH \tag{3}$$

Following this step, acidogenic bacteria convert the soluble monomers, such as sugars, amino acids, and fatty acids, into longer-chain VFAs such as acetate and butyrate (Eqs. 4 and 5), along with other byproducts such as H_2, CO_2, ethanol, and NH_3 [38].

$$C_6H_{12}O_6 \rightarrow 2 \quad CH_3COOH + 2CO_2 + 4H_2 \tag{4}$$

$$C_6H_{12}O_6 \rightarrow CH_3CH_2CH_2COO^- \quad + \quad 2CO_2 + 2H_2In \tag{5}$$

the acetogenesis stage, longer-chain VFAs (like propionate and butyrate) are further oxidised into acetic acid, CO_2, and H_2 by syntrophic acetogenic bacteria under low hydrogen pressure.

$$CH_3CH_2CH_2COO^- + \quad 2H_2O \rightarrow 2CH_3COOH + 2H_2 \tag{6}$$

The products of acidogenic and acetogenic fermentation serve as substrates for methanogenic microorganisms in the later stages of AD, which produce biogas [37,39]. Organic residues, including food waste, agro-industrial residues, and municipal solid waste (MSW), can be converted into VFAs through anaerobic digestion and have shown high conversion efficiencies in currently available literature, as demonstrated in Table 4. During anaerobic digestion, the type of waste digested strongly influences the VFA yield and composition [37,40,41]. Strazzera et al. (2021) demonstrate this in their study, showcasing that when fermenting food waste rich in proteins, the VFA profile is typically characterised by acetic acid as the main VFA product, accounting for around 70% of the total VFAs produced, while butyrate and propionate represent around 10% and 15% of the total VFAs, respectively [42]. However, other VFAs, such as propionic and valeric acids, can also be significant, depending on the specific proteins and digestion conditions. Factors such as fermentation pH, temperature, and organic loading can influence the relative distribution of VFAs, leading to different

Table 4
Studies exploring VFA production using various waste streams.

Feedstock / Waste Type	Treatment Method	Operating Conditions	VFA Composition (Major Acids)	Total VFA Concentration (mg/L)	Normalised VFA Yield (g VFA/g VS feed)	Ref
OFMSW* & food waste	Mesophilic acidogenic fermentation	T = 35 °C, Time = 3.5 d, pH = 6.0	Acetic, propionic, and butyric	~11,500	0.45–0.55	[71]
Food waste (Tuna)	Anaerobic digestion	T = 30℃, Time = 20 d, pH = 5	acetic, propionic, butyric and valeric acids	4060	~0.73	[41]
Slaughterhouse wastewater	Anaerobic digestion (alkaline fermentation)	T = 35℃, Time = 10 d, pH = 10	Acetic, propionic, and butyric	3357	0.611–0.67	[45]
Food waste	Anaerobic digestion/ Acidogenic fermentation	T = 35℃, Time = 5 d, pH = 5	Acetic, propionic, and butyric	34,000	~0.53	[58]
Pulp and paper mill	Anaerobic digestion	T = 39℃, Time = 72 h, pH = 7	Acetic, propionic, and butyric	3580	–	[72]
Spent coffee grounds	Hydrothermal liquefaction (using nitrogen)	T = 300℃, Time = 30 min, P = 85 bar	Acetic, formic, propionic	~836	–	[52]
Polymers	Hydrothermal deconstruction (using oxygen)	T = 280℃, Time = 90 min, $P_{Initial}$ = 30 bar	Acetic acid	8974	–	[69]
MSW	Hydrothermal deconstruction (using oxygen)	T = 280℃, Time = 45 min, $P_{Initial}$ = 35 bar	Acetic acid	2930	–	[68]
Olive mill solid waste	Anaerobic fermentation (hydrothermal pretreated)	T = 35 °C, Time = 15 d, pH = 8	Isobutyric, butyric, and valeric	~7000	0.25–0.30	[73]

* OFMSW refers to the organic Fraction of Municipal Solid Waste

outcomes; some studies show high proportions of valeric acid alongside acetic acid from protein-rich substrates [38,39]. This variation in resulting VFAs is largely determined by the operational and environmental conditions maintained during anaerobic digestion. Among these, pH plays a pivotal role as it directly affects the activity and stability of acidogenic microorganisms [43]. Mildly acidic to neutral conditions (pH 5.5–6.5) generally favour acidogenic bacteria that convert soluble organics into volatile fatty acids (VFAs), whereas more acidic environments (<5.5) can inhibit microbial enzymes and slow down hydrolysis [43,44]. In contrast, strongly alkaline conditions (pH 9–10) have been shown to enhance VFA accumulation rather than suppress it [45,46]. Under such conditions, methanogenic archaea are effectively inhibited, preventing VFA consumption, while alkaliphilic fermentative bacteria such as *Clostridium* and *Bacillus* spp. remain active. The high pH also promotes the solubilization of complex organic matter, improving substrate availability for acidogenesis [47]. As fermentation progresses, the pH typically decreases due to acid production, creating a self-regulating environment that sustains acidogenic activity and supports elevated VFA yields [45,46,48]. Additionally, temperature significantly influences VFA production by affecting reaction kinetics and microbial community structure. Thermophilic conditions (~55°C) are known to enhance enzymatic hydrolysis and acidogenesis rates, accelerating organic matter breakdown and VFA formation. However, this can also destabilise microbial communities if not carefully controlled, leading to increased inhibitory ammonia formation and posing stability risks for microbial cultures. Mesophilic conditions (~35°C) are more common for continuous AD systems, especially when handling high-solids waste inputs, as they form more stable conditions for microbes during fermentation. However, they are observed to typically produce VFAs at slower rates [49,50].

2.1.2. *VFA production through hydrothermal liquefaction (HTL)*

Thermochemical processes such as hydrothermal liquefaction (HTL) are typically conducted under supercritical to subcritical water conditions (250–500 °C and 100–300 bar) in a non-oxidative environment, creating a reducing or anoxic reaction environment [51,52]. Under these conditions, water acts simultaneously as a reactant, solvent, and catalyst, with an elevated ionic product (K_w) enhancing acid–base catalysed hydrolysis and solvolysis reactions. These conditions favour bond cleavage and depolymerisation, but also promote secondary recombination and condensation, meaning that VFAs are generally transient intermediates rather than final products in HTL [51]. Commonly, short-chain organic acids are favoured under moderate HTL severities (~280–300 °C and residence times of ~5–30 min), where hydrolysis and fragmentation rates exceed recombination kinetics [52]. At higher temperatures (>350 °C) or longer residence times (>30 min), VFAs are increasingly consumed via decarboxylation, aldol condensation, and aromatisation reactions, contributing to the formation of biocrude and gaseous products [52,53]. The absence of molecular oxygen further limits the stabilisation of carboxylic acids, favouring reduced products and suppressing net VFA accumulation [54,55].

Protein-rich feedstocks readily hydrolyse of peptide bonds under subcritical water conditions, yielding free amino acids. These amino acids subsequently decompose via deamination and decarboxylation, yielding ammonia, CO_2, and short-chain organic acids. Acetic and propionic acids are the dominant VFAs formed, depending on amino acid side-chain structure, as shown in Eq. 7 [56,57].

$$RCH(NH_2)COOH + H_2O \rightarrow CH_3COOH + NH_3 + CO_2 \quad (7)$$

This pathway explains the consistent observation of VFAs as intermediates in HTL of protein-rich feedstocks such as sewage sludge, food waste, and spent coffee grounds. For example, Liakos et al. (2025) reported peak concentrations of acetic (540.7 mg L^{-1}), propionic (155.6 mg L^{-1}), and formic acids (67.8 mg L^{-1}) at 300 °C during HTL of spent coffee grounds, indicating an optimal balance between hydrolysis and secondary degradation at this temperature [52]. While Liakos et al. (2025) identify 300 °C as the optimal balance for VFA recovery from coffee grounds, the reported concentrations highlight a critical limitation of HTL compared to biological routes. In contrast to anaerobic digestion, which can yield acetic acid concentrations exceeding 34,000 mg/L from food waste, the low steady-state concentrations in HTL suggest that high secondary degradation rates significantly undermine its viability as a primary VFA source [58]. This suggests that HTL may be better suited as a rapid pretreatment step to enhance feedstock solubilization rather than as a standalone VFA production method.

Alternatively, polysaccharides such as cellulose and starch undergo rapid hydrolytic depolymerisation to monomeric sugars under HTL conditions (Eq. 8).

$$C_6H_{10}O_5 + H_2O \rightarrow C_6H_{12}O_6 \quad (8)$$

Monosaccharides then decompose through retro-aldol cleavage, dehydration, and fragmentation, forming lactic acid, formic acid, and acetic acid as dominant VFAs. Lactic acid is often a key intermediate, subsequently decomposing into acetic acid, CO_2, and H_2 (Eqs. 9 and 10)

[52].

$$C_6H_{12}O_6 \rightarrow 2CH_3COOH + CO_2 + H_2 \quad (9)$$

$$C_3H_6O_3 \rightarrow CH_3COOH + CO_2 + H_2 \quad (10)$$

Although carbohydrates readily generate VFAs, these acids are unstable under prolonged HTL conditions and frequently undergo secondary condensation reactions, contributing to phenolic and aromatic structures in the biocrude phase [52]. This explains the typically lower steady-state VFA concentrations observed in carbohydrate HTL compared with those in oxidative thermochemical processes. Conversely, when processing more complex organic waste structures such as plastics (Fig. 2), the degradation pathways in HTL depend strongly on polymer chemistry and the presence of reactive sites [51,59,60]. Studies suggest that, unless co-liquefaction of polymers mixed with oxygen-rich feedstocks is performed, no oxygenated intermediates or VFAs are formed [51,61]. The relatively low steady-state VFA concentrations and high secondary degradation rates suggest that HTL-derived streams may require further optimisation or integration before being viable substrates for efficient PHA production.

2.1.3. VFA production through hydrothermal deconstruction (HTD)

In contrast to HTL, hydrothermal deconstruction (HTD), often referred to as wet air oxidation (WAO), introduces oxidants (air, O_2, or H_2O_2) under 200–350 °C and 20–200 bar [51,62]. Under HTD/WAO conditions, oxidation proceeds primarily through radical-chain pathways, which tend to fragment complex organics into oxygenated intermediates [63]. In partial oxidation regimes, the dominant stable intermediates are often short-chain carboxylic acids, with acetic acid repeatedly identified as a major partial oxidation product of HTD [51,62].

Protein-rich waste streams hydrolyse to amino acids, followed by oxidative deamination and decarboxylation reactions that generate NH_3, CO_2, and low-molecular-weight oxygenates, including organic acids (Eq. 11) [64,65]. Similarly, carbohydrate-rich waste streams undergo hydrolysis to sugars, then oxidative fragmentation, yielding acetic and other small-chain acids (Eq. 12) [64]. In wet oxidation, aromatic/phenolic structures (e.g., lignin-derived) are progressively opened and oxidised through oxygenated intermediates, with acetic acid commonly appearing as a key partial oxidation product.

$$RCH(NH_2)COOH + O_2 \xrightarrow{H_2O} CO_2 + H_2O + \quad CH_3COOH + NH_3 \quad (11)$$

$$C_6H_{12}O_6 \quad + 2O_2 \xrightarrow{H_2O} 2CO_2 + 2H_2O + \quad 2CH_3COOH \quad (12)$$

A major difference compared to VFA production via HTL is that HTD/WAO can oxidatively break complex structures, including polymer-derived fragments, into oxygenated products even when the polymer lacks hydrolysable bonds (Fig. 3), though the extent and selectivity depend on oxygen availability and severity [51,66]. Polyesters and polyolefins such as PET and PP undergo oxidative cleavage to oxygenated intermediates and acids. Studies treating polymer-containing wastes (including PET- and PP- containing feedstocks) using HTD methods report strong solids reduction and high acetic acid concentrations when oxidation severity is tuned to stop at partial oxidation [67].

$$n(C_2H_4) + O_2 \xrightarrow{H_2O} CO_2 + H_2O + \quad CH_3COOH \quad (13)$$

In contrast, polyamides hydrolyse more slowly than polyesters but can be oxidatively deconstructed under HTD conditions, with formation of oxygenated intermediates and nitrogen-containing by-products (e.g., ammonia) that can persist unless severity/catalysis is increased (Eq. 14) [51,67].

$$\text{Polyamides} + O_2 \xrightarrow{H_2O} CO_2 + H_2O + CH_3COOH + NH_3 \quad (14)$$

Anthraper et al. (2018) explored the use of HTD to treat non-recyclable solid waste materials found in municipal solid waste (MSW). They found that at around 280°C, over 90% of the solids were reduced, and a high concentration (up to 2930 mg L^{-1}) of acetic acid was recovered [68]. Significantly higher VFA levels were observed by Javid et al. (2022), recovering up to 8974 mg L^{-1} of acetic acid from a personal protective equipment (PPE) mixture, including safety goggles, gowns, and gloves [69]. This method is particularly valuable for heterogeneous waste streams, such as those containing polymeric materials, where biological degradation is limited.

However, a major limitation lies in the presence of additives, inorganics, and hazardous by-products such as pigments, flame retardants, stabilisers, and fillers as they may co-extract with VFAs during HTD, change solution chemistry, and interfere with separation, purification, and biological upgrading [70]. The disparity in VFA yields between MSW and PPE mixtures under similar HTD conditions reveals an inconsistency that depends heavily on the initial polymer's oxygen content. The significantly higher yields from PPE likely stem from the oxidative cleavage of oxygen-rich polymer chains (e.g., polyamides/polyesters), which are more susceptible to radical-chain fragmentation than the heterogeneous organic matter in MSW [69]. While HTD achieves impressive solids reduction (90%), the method's effective VFA production is limited by a trade-off between oxidation severity and acid stability as excessive severity risks over-oxidation to CO_2, while

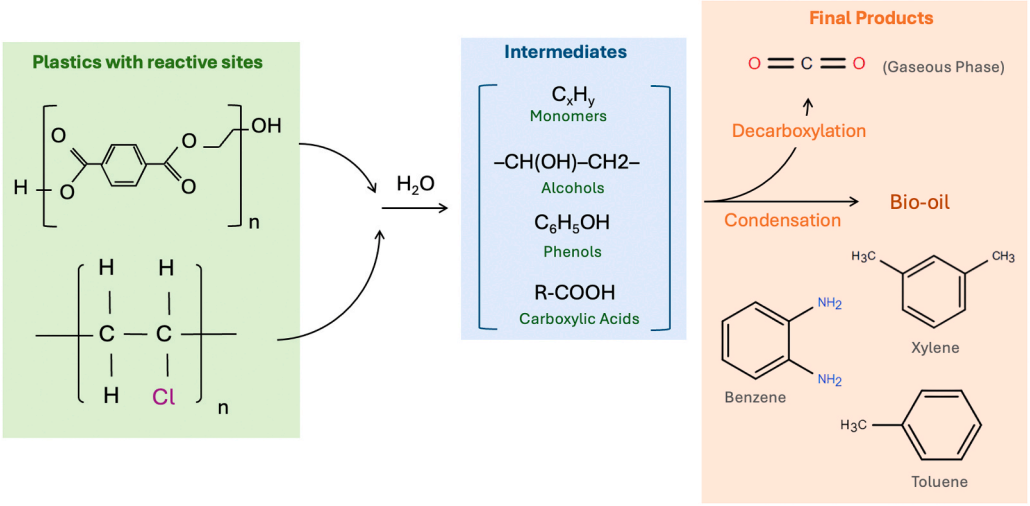


Fig. 2. HTL mechanism for polymers with reactive sites (e.g., PET and PVC).

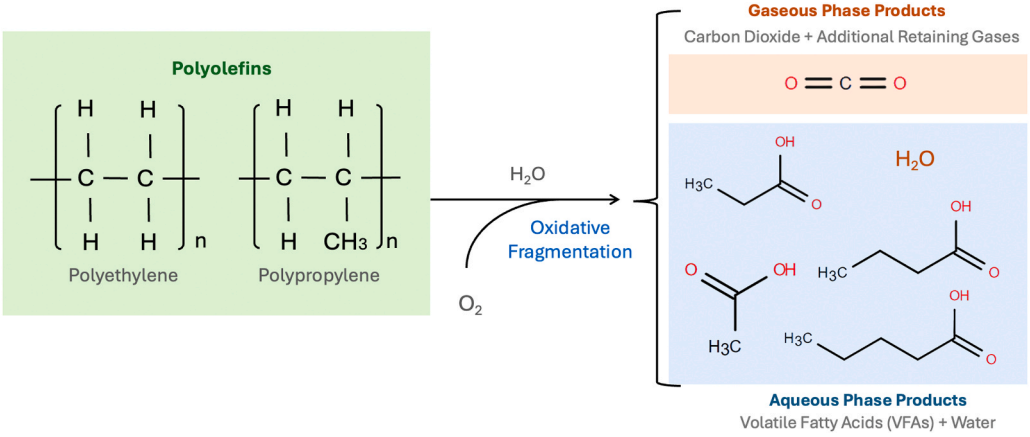


Fig. 3. HTD mechanism for plastics (e.g., polyolefins).

insufficient severity leaves complex polymers untouched [70]. Importantly, the presence of oxidation-derived impurities and additives in HTD liquors introduces additional challenges for downstream microbial fermentation, necessitating careful consideration for PHA applications. Table 4 summarises VFA production across different waste streams, highlighting parameters that are particularly relevant for downstream PHA synthesis, including acid composition, concentration, and process-dependent variability.

2.2. VFA separation and purification

Despite promising yields, VFAs recovered from organic waste streams are chemically challenging to separate due to their intrinsic properties. They are short, low-molecular-weight carboxylic acids with pKa values around ~3.75–4.9 [74,75]. As a result, under typical neutral/alkaline process conditions, they exist mainly as highly water-soluble, dissociated carboxylate anions rather than as volatile free acids [75]. This high water solubility, combined with relatively similar molecular weights and overlapping boiling points for C2–C5 acids, reduces volatility and selective separability by simple distillation and makes selective extraction difficult without pH adjustment or specialised unit operations [76,77]. Table 5 summarises the VFA separation and purification techniques commonly used for the pretreatment of VFA-rich liquors in industry.

2.2.1. Pertraction (membrane-assisted solvent extraction)

Pertraction is a membrane-assisted solvent extraction method where, instead of mixing the fermentation broth directly with an organic solvent, which can form stable emulsions that are hard to separate, the two phases are separated by a hydrophobic microporous membrane [79]. During this process, the VFA molecules diffuse from the aqueous feed through the membrane pores and are extracted into the solvent on the other side [79]. Separation is typically performed under acidic conditions (pH ~2–4) to maintain the acids in their undissociated, hydrophobic form, promoting transfer into the organic phase. A pH gradient is commonly applied, with an alkaline stripping phase to back-extract and concentrate the recovered acids. Under these conditions, recoveries of ~70–95% have been reported [78,79]. While this process successfully avoids emulsion formation during solvent separation, the membrane introduces mass-transfer resistance, which can slow the process compared to direct extraction [79]. Furthermore, transport generally increases with VFA hydrophobicity, meaning medium-chain acids are recovered more effectively. Similar to most membrane technologies, long-term performance may be limited by membrane wetting and fouling by suspended solids or organic matter in complex broths [79].

Table 5
Summary of VFA separation and purification techniques commonly used for the pretreatment of VFA-rich liquors.

Technique	Efficiency / Performance*	Optimal Operating Conditions	Effectively extracted VFAs	Ref.
Pertraction	High (~70–95%)	Acidic pH and moderate to ambient temperatures.	Butyrate and valerate are preferred (higher hydrophobicity improves flux).	[78, 79]
Pervaporation	Moderate–High	Acidic pH, vacuum permeate, and Temperatures between 30–60 C.	Mainly acetic/ propionic acids. Small acids with high volatility are preferred.	[80, 81]
Reactive extraction	Very high (~80–99%)	Acidic pH, amine-based extract, and alkaline stripping for regeneration.	Broad range (C2–C6). Particularly effective for hydrophobic VFAs.	[82, 83]
In-situ product removal (ISPR)	Indirect**	Mild temperatures, continuous extraction and absorption loop.	All inhibitory VFAs.	[84, 85]
Electrodialysis with bipolar membranes (EDBM)	High (~80–95%)	Neutral–alkaline pH, VFAs in carboxylate form, and sufficient conductivity.	All dissociated VFAs.	[86, 87]
Ion-exchange resins/ adsorption	Moderate–High	Neutral–alkaline pH, VFAs in carboxylate form.	Long-chain acids show stronger hydrophobic adsorption.	[88, 89]

* Performance depends strongly on acid chain length, pH, ionic strength, membrane/resin type, and feed composition.
** Improves productivity rather than separation.

2.2.2. Pervaporation

Pervaporation is a membrane process that utilises selective polymeric or inorganic membranes to separate VFAs from a liquid mixture [81]. The volatile compound-rich broth is placed in contact with a selective membrane, often hydrophobic. The VFAs selectively absorb into the membrane, diffuse across it, and evaporate on the other side into a vacuum or sweep gas stream [80,81]. The driving force for this separation procedure is the difference in the partial pressures of the VFA

mixture across the membrane. Operation is typically most effective under acidic conditions (undissociated acids) and at moderate temperatures (~30–60 °C) to enhance permeability while preserving membrane integrity. When optimised, the process may achieve moderate to high removal efficiencies (~50–90%). Pervaporation is more effective for short-chain, more volatile acids such as acetic and propionic acids, and is frequently applied for concentration or polishing rather than bulk recovery. This process consumes less energy than distillation, as only the permeating fraction of VFAs is vaporised [81]. However, the flux rates of this process are relatively low, requiring a large membrane surface for larger-scale operation. Additionally, pervaporation poses long-term challenges, including membrane fouling, degradation over prolonged use, and selectivity issues when dealing with complex mixtures [81].

2.2.3. Reactive extraction

Reactive extraction using tertiary amines or ionic liquids is a liquid-liquid separation process that uses a chemically reactive solvent to selectively extract VFA from aqueous streams [82,83]. Tertiary amines act as bases, forming reversible acid-amine complexes that partition preferentially into the organic phase [82]. This method is generally conducted at acidic pH to maximise the extraction of undissociated acids, resulting in high recoveries (~80–99%) and making the process particularly effective for dilute broths. Extraction efficiencies generally increase with carbon chain length. As a result, propionate, butyrate, and valerate are often recovered more readily than acetate [90]. While the process substantially reduces solvent requirements compared to physical extraction, regeneration of the solvent and recovery of pure acids require bac-extraction or alkaline stripping, increasing chemical consumption and process complexity [82].

2.2.4. In-situ product removal (ISPR)

ISPR strategies remove VFAs as they are produced (e.g., via adsorption, membrane extraction, or solvent extraction integrated into the bioreactor), helping to avoid product inhibition, shift reaction equilibria, and increase overall productivity [84,85]. By continuously withdrawing inhibitory acids, ISPR can reduce downstream load and energy needs by capturing acids early. These systems typically operate at mild temperatures and under biocompatible conditions, requiring selective materials that do not impair microbial activity. This technology can be applied broadly to all VFA species, depending on the coupled separation technology. However, it requires reactor integration, compatible removal technologies that don't harm microbes (e.g., non-toxic adsorbents or membranes), and robust control to handle variable production rates [85].

2.2.5. Electrodialysis with bipolar membranes (EDBM)

The EDBM process splits ionic VFAs into their acid and base forms by applying an electrical potential across stacks of ion-exchange membranes, producing a concentrated acid stream and a corresponding base (e.g., NaOH) [86,87]. This process works directly on dissociated acids and avoids the use of organic solvents, making it effective for recovering VFAs from dilute, saline streams, making it most effective at neutral to alkaline conditions [86]. Under these conditions, recoveries of 80–95% are recorded. While effective, EDBM is generally not selective among different VFAs, and major challenges include significant electrical energy demand, membrane fouling in complex waste matrices, and the need to pre-remove particulates and foulants to protect membranes [86,87].

2.2.6. Ion-exchange resins and adsorption

Ion-exchange resins (strong or weak base resins used in the acid form) and other adsorbents can capture VFA carboxylates from aqueous streams and later regenerate them to recover concentrated acids [88]. These systems operate most effectively at neutral or alkaline pH, where VFAs are present as carboxylate anions, and are particularly suited to dilute streams that require pre-concentration. Typical capture efficiencies range from 60 to 95%, and substantial concentration factors can be achieved upon elution. Short-chain acids such as acetate and propionate are commonly recovered, although adsorption strength generally increases with hydrophobicity. Challenges include competition from other anions (such as salts, sulphate, and phosphate), resin fouling, regeneration requirements (including chemical and salt management), and resin degradation over time, all of which affect operating costs and lifecycle sustainability [88,89].

Consequently, the most suitable VFA production route for PHA synthesis cannot be judged on acid yield alone. The downstream value of the liquor depends on a combination of factors, including VFA composition, concentration, impurity profile, ease of recovery, and reproducibility across waste batches. These characteristics ultimately determine whether the recovered VFAs can be used directly for fermentation, require polishing before use, or impose process-control and purification costs that challenge large-scale implementation.

3. Utilising VFAs for PHA biosynthesis

The utilisation of economically viable substrates, such as VFAs, especially those derived from readily available waste streams, may significantly reduce costs and facilitate the production of final PHA products [13]. Waste-derived VFAs are considered preferred carbon sources for PHA synthesis, as they serve as direct metabolic precursors during PHA production.

3.1. Metabolic pathways of VFA-to-PHA conversion

Depending on the carbon feedstock supplied during PHA synthesis, different intracellular metabolic pathways are activated. When VFAs serve as the primary carbon source, bacterial cultures such as *Cupriavidus necator* uptake VFAs (e.g., acetic, propionic, butyric, or valeric acids) either by passive diffusion, in undissociated form, or via specific membrane transport proteins that regulate movement across the lipid bilayer [22,25,91]. Once inside the cell, VFAs are rapidly activated into their corresponding coenzyme A (CoA) derivatives, such as acetyl-CoA from acetate and propionyl-CoA from propionate [92,93]. These activated intermediates act as central metabolic nodes, linking carbon assimilation to energy generation, lipid metabolism, and PHA biosynthesis [13]. During synthesis, three main metabolic pathways lead to PHA production. Pathway I (acetoacetyl-CoA route) primarily yields short-chain-length PHAs (scl-PHAs), where two acetyl-CoA molecules are first condensed by β-ketothiolase (PhaA) to form acetoacetyl-CoA [94]. This intermediate is then stereospecifically reduced by acetoacetyl-CoA reductase (PhaB) to (R)-3-hydroxybutyryl-CoA, which serves as the direct monomer substrate for PHA synthase (PhaC). Enzyme PhaC catalyses the polymerisation of these monomers into intracellular PHB granules [13]. This enzyme set (PhaA–PhaB–PhaC) is characteristic of scl-PHA biosynthesis, especially when acetate is the dominant substrate [94]. Alternatively, in pathway II (de novo fatty acid synthesis), acetyl-CoA enters the fatty acid biosynthetic cycle, where acyl-ACP intermediates are generated; these are subsequently transferred to CoA-bound hydroxyacyl-CoA monomers via enzymes such as PhaG (3-hydroxyacyl-ACP:CoA transferase) before being polymerised by PhaC [24]. This route contributes mainly to mcl-PHAs and selected copolymer structures, depending on the chain length of the diverted intermediates and the availability of odd-carbon precursors [94].

Pathway III (fatty acid β-oxidation) is mainly utilised when longer-chain VFAs are supplied. Partial β-oxidation cycles generate hydroxyacyl-CoA intermediates that are directly incorporated into medium-chain-length PHAs (mcl-PHAs) such as PHHx or PHO [94]. Pathway III uses exogenous fatty acids, where the partial β-oxidation cycle generates enoyl-CoA (via enoyl-CoA hydratases such as PhaJ) and (R)-3-hydroxyacyl-CoA intermediates that are directly channelled by PhaC to mcl-PHA monomers [94]. Commonly, odd-chain VFAs (e.g. propionate) can also be converted to propionyl-CoA and incorporated as

3-hydroxyvalerate (3HV) comonomers, enabling PHBV production. These three routes (Fig. 4) explain how the same VFA feedstock composition can yield different PHA monomer distributions depending on which pathway(s) predominate under the chosen microbial/operational conditions [13,92].

The type of VFA supplied strongly governs the resulting polymer composition. Even-carbon VFAs such as acetate predominantly yield PHB units, whereas odd-carbon VFAs such as propionate and valerate generate propionyl-CoA intermediates that enable the incorporation of 3HV monomers and the formation of copolymers such as PHBV [22,25,91]. Furthermore, under nutrient-limiting conditions, particularly nitrogen and phosphorus, bacterial growth is restricted, and excess carbon is redirected toward intracellular PHA accumulation as storage granules, which can subsequently be recovered and purified for bioplastic applications.

3.2. Microbial cultures used for PHA synthesis

A range of microbial systems has been investigated for PHA synthesis from waste-derived VFAs, including pure microbial cultures, haloarchaea, and mixed microbial cultures (MMCs). To provide a clearer overview, Table 6 summarises the principal microorganisms reported in the literature together with the type of PHA produced, the VFA substrate used, the culture mode, key operating conditions, and the reported PHA accumulation. Pure cultures offer better process control and polymer specificity, whereas MMCs are generally more robust toward variable waste-derived substrates and may be more attractive for large-scale implementation.

Pure cultures enable genetic optimisation and metabolic engineering, which can yield tailored polymer properties but often result in lower PHA yields. However, they provide better process control and predictability, since only one strain with known metabolism is used [95]. Notably, the bacterial strain *Ralstonia eutropha* (alternatively known as *Cupriavidus necator*) showed the most variation in PHA content, with reported yields ranging from ~30–80% in dry cell weight (DCW) as demonstrated in Table 6 [96]. Hernández-Herreros et al. (2025) observed an optimal accumulation of PHB biopolymers of ~75% DCW with a pure culture of *Ralstonia eutropha* at 30°C, pH of 6.8, 20% dissolved oxygen (DO) level, and 6 g/L VFA feed concentration [97]. This bacterial strain can efficiently utilise a variety of VFAs, such as acetic, propionic, and butyric acids, to produce a favourable amount of PHAs [96]. These results are comparable to those reported by Yun et al. (2013), who utilised *Ralstonia eutropha* strains and achieved ~50% DCW of PHB from a mixture of various VFAs under optimal fermentation conditions (30°C, 6.5 pH, and 20% DO) [96]. The significant increase in accumulation from 50% in batch to 75% in fed-batch for *Cupriavidus necator* suggests that substrate-to-PHA conversion is not limited by the microbe's metabolic capacity, but rather by the inherent toxicity of VFAs at high concentrations. This confirms that the shortened metabolic pathway of VFAs provides a competitive advantage only if toxicity is strictly managed through feeding strategies.

In contrast, *Haloferax mediterranei*, a halophilic archaeon, has been studied for PHBV production using VFAs as carbon substrates. Ferre-Guell and Winterburn (2019) investigated the production of PHBV biopolymers using a *Haloferax mediterranei* with a mixture of butyric and valeric acids as the main carbon sources. They found that at around 37°C, pH of 6.8, and 20% DO, the highest PHBV content reached was ~60% by DCW [98]. The significant advantage of *Haloferax mediterranei* species lies in its ability to grow and produce PHAs in non-sterile, saline environments, thereby reducing PHA production costs and, in some

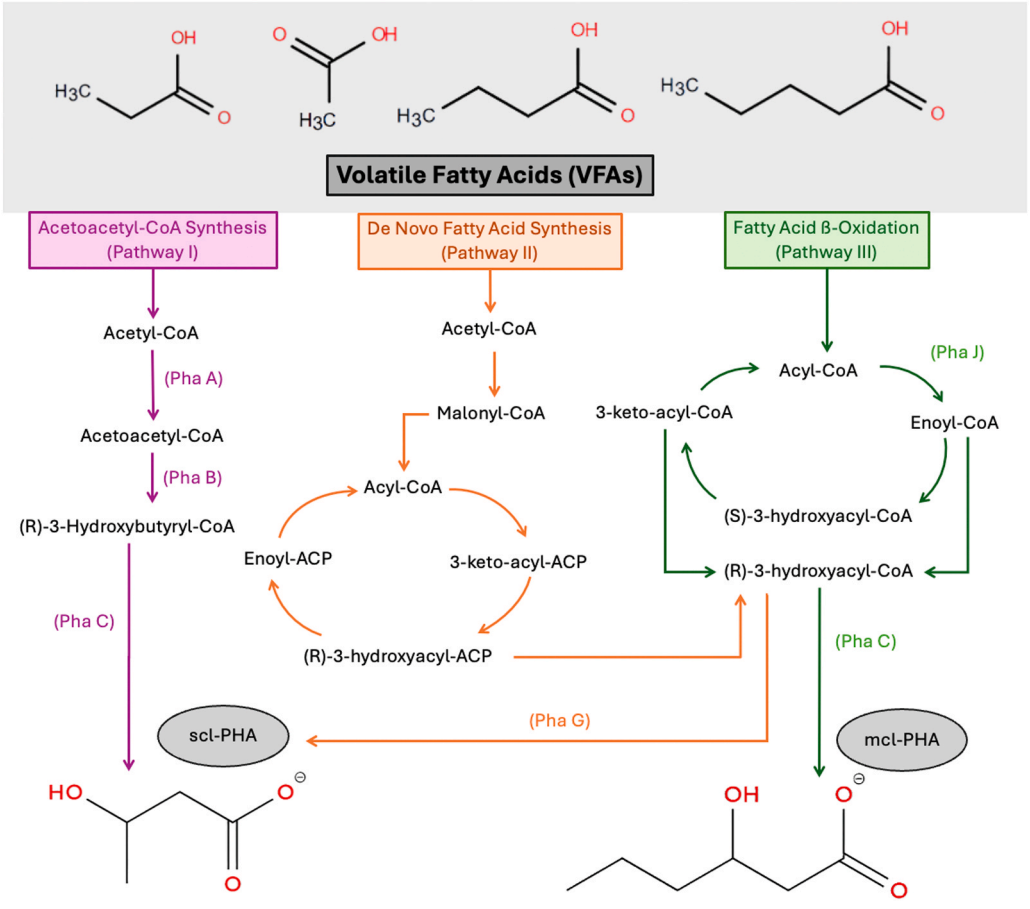


Fig. 4. Metabolic pathways of PHA production using VFAs as carbon substrates.

Table 6
Microbial cultures used for PHA production from waste-derived VFAs.

Microorganisms	Type of PHA produced	VFA carbon source	Culture Mode	Culture Conditions	PHA content (%DCW)	Key Advantage	Ref.
Ralstonia eutropha / Cupriavidus necator	PHB and PHBV	Acetic acid, propionic acid, butyric acid	Batch Fermentation	T = 30°C pH = 6.5 DO = 20% Feed concentration = 5 g/L	~50		[96]
	PHB	Acetic acid, propionic acid, butyric acid, and valeric acid	Fed Batch Fermentation	T = 30°C pH = 7.4 DO = 20% Feed concentration = 6 g/L	~75	High accumulation; well-established model strain	[97]
	mcl-PHA, PHB, and PHBV	Acetic and propionic acid	Batch Fermentation	T = 30°C pH = 7.5 DO = 20% Feed concentration = 2.5 g/L	~53		[106]
Haloferax mediterranei	PHBV	Butyric acid, and valeric acid	Fed-batch fermentation	T = 37°C pH = 6.8 DO = 20% Feed concentration = 4.25 g/L	59		[98]
	PHBV	Acetic acid, propionic acid, and butyric acid	N/A	T = 37°C pH = 7 Feed concentration = 4 g/L C/N ratio = 5 C/P ratio = 35	55	Non-sterile saline cultivation; reduced sterilisation demand	[107]
	PHBV	Butyric acid and valeric acid	Continuous Fermentation	T = 37°C pH = 6.8 DO = 20% Feed concentration = ~ 4 g/L	~60		[108]
Rhodococcus sp.	PHBV	Acetic acid, valeric acid	Batch Fermentation	T = 30°C pH = 7 Feed concentration = 5 g/L	~30	Better incorporation of 3HV; useful for flexible PHBV	[99]
	PHB and PHBV	Acetic acid, propionic acid, and butyric acid	Batch Fermentation	T = 30°C pH = 7 Feed concentration = 2.2 g/L	~48		[109]
Bacillus sp.	PHB and PHBV	Propionic acid and glucose	Batch fermentation	T = 37°C pH = 7 DO = 20% Feed concentration = 1.5 g/L	~33	Robust strain; attractive for food/biomedical relevance	[101]
	PHBV	Glucose, acetic acid, and propionic acid	Fed-batch Fermentation	T = 30°C pH = 7 DO = 4.1 mg/L Feed concentration = 1 g/L	25		[102]
Pseudomonas putida / Pseudomonas sp.	PHBV	Glucose, acetic acid, and propionic acid	Fed-batch Fermentation	T = 30°C pH = 7 DO = 4.1 mg/L Feed concentration = 1 g/L	35	Suitable for mcl-PHA production	[102]
	mcl-PHA	Acetic acid, propionic acid, butyric acid, and valeric acid	Fed-batch Fermentation	T = 30°C pH = 6.8 Feed concentration = 3.4 g/L	~45		[103]
Mixed microbial culture (MMC)	PHB	Pure acetate	Fed-batch fermentation	T = 30°C pH = 7 Feed concentration = 3.8 g/L	~89	High substrate flexibility; suitable for variable waste-derived feeds	[104]
	PHBV	acetate, propionate, butyrate and valerate	Two-stage fermentation	T = 23 – 25°C pH = 8 Feed concentration = ~1 g/L	~77		[105]

cases, achieving higher PHA contents than most alternative pure culture PHA producers.

Another strain known to support VFAs as carbon sources for PHA production is *Rhodococcus sp.* In their study, Haywood et al. (1991) reported that *Rhodococcus sp.* can incorporate valeric acid into the polymer backbone, enabling the synthesis of PHBV copolymers with higher 3HV fractions compared to *Cupriavidus necator* [99]. The higher 3HV fractions are known to modify the thermal and mechanical properties of the polymer, resulting in a less brittle material more suitable for flexible applications [100]. Their research indicated that at favourable

conditions (30°C, and pH of 7), up to ~30% DCW was obtained using a mixture of acetic and valeric acids as main carbon substrates [99]. Additionally, in recent literature, *Bacillus* species have been identified as promising PHA producers due to their robustness and ability to secrete extracellular enzymes [101]. Several Bacillus strains have been reported to accumulate PHAs such as PHB and co-polymers PHBV of up to ~20–30% DCW at temperatures ranging between 30–37 °C, neutral pH of 7, a DO content of 20%, and feed concentrations between 1–1.5 g/L [101,102]. The *Bacillus* strains show the greatest efficiency for PHA production when VFAs are combined with glucose as carbon sources. A unique advantage of *Bacillus* strains lies in their recognised safe status, making them attractive for industrial-scale production, where downstream processing might involve food or biomedical applications [101, 102]. Conversely, certain bacterial species have the ability to produce mcl-PHAs, enabling their use in many industries where scl-PHAs may fall short. *Pseudomonas putida* and related *Pseudomonas* species are known for their ability to utilise VFAs for the production of mcl-PHAs. These biopolymers differ from the scl-PHAs produced by other bacterial strains discussed, as they result in polymers with higher elastomeric properties, higher flexibility, and lower crystallinity, making them particularly suitable for biomedical applications. Reported yields of accumulated mcl-PHAs within *Pseudomonas* strains range from ~20–50% DCW at 30 °C and pH between 6.8–7 [102,103]. The inconsistency in yields across studies using the same bacterial species further underscores that VFA-to-PHA conversion is more sensitive to cultivation mode and inhibitor load than it is with traditional sugar-based substrates. Furthermore, *Pseudomonas* strains can metabolise a broad range of feedstocks, including fatty acids, plant oils, and even aromatic compounds, highlighting their strong potential in waste valorisation processes.

While pure bacterial cultures may lead to more tailored PHA properties, they require strict sterile conditions and refined feedstocks, which further increase production costs of PHAs. Instead, research on utilising mixed microbial cultures (MMC) has been advancing further due to lower associated costs, higher substrate flexibility, and better suitability for waste-based, large-scale PHA production. When cultured under optimal fermentation conditions, they often yield higher PHA accumulation than pure microbial cultures. For instance, Johnson et al. (2009) investigated the production of PHAs using MMC cultures, reporting the maximum PHA content of 89% DCW using pure acetate at 30°C and a neutral culture medium (pH of 7) [104]. Similarly, Albuquerque et al. (2011) obtained maximum PHA contents between 56% and 77% DCW with the VFA mixtures from fermented molasses feed and synthetic feedstocks, respectively [105]. These PHA fermentation trials were performed at temperatures ranging from 23 to 25°C and a slightly alkaline culture medium (pH of 8). The utilisation of MMCs represents a major technical bottleneck for material consistency, however. While MMCs are robust to the compositional variability of waste, they inherently lack the metabolic precision to produce the high-purity, specialised copolymers (e.g., mcl-PHAs) required for medical-grade applications.

Overall, the literature indicates that no single microbial platform is universally optimal for all waste-derived VFA streams. *Cupriavidus necator* remains one of the most effective and widely studied strains for high PHA accumulation, particularly under controlled fed-batch operation, whereas *Haloferax mediterranei* offers practical advantages under non-sterile saline conditions. Pseudomonas species are especially relevant when mcl-PHAs are desired, while MMCs are attractive for variable waste streams because of their operational robustness and high accumulation potential. Accordingly, strain selection should be matched not only to VFA composition and inhibitor load, but also to the desired polymer type and the intended scale of production.

3.3. Parameters influencing PHA growth & properties

Mixed VFA streams derived from heterogeneous feedstocks, such as municipal waste streams, along with the processing parameters (summarised in Table 7) used to extract these VFAs, directly determine the composition and properties of the resulting PHA.

Different VFAs are metabolised into distinct hydroxyacyl-CoA precursors, which are incorporated into the growing polymer chain, offering adjustable mechanical and thermal properties through controlled monomer incorporation. When acetate is the dominant substrate, it is primarily converted into 3-hydroxybutyrate (3HB) units via acetyl-CoA, leading to the formation of the highly crystalline, stiff, and brittle PHB homopolymer [110,111]. In contrast, feeding odd-chain VFAs such as propionate and valerate leads to the intracellular formation of propionyl-CoA and valeryl-CoA, which are converted into 3HV precursors and subsequently incorporated into the polymer backbone [116]. As the fraction of 3HV increases, the polymer shifts from PHB toward the copolymer PHBV. According to the literature, increasing the 3HV content disrupts the polymer chain packing, reduces crystallinity and the melting temperature, decreases stiffness and brittleness, and increases ductility, impact resistance, and toughness [117–119]. Consequently, higher propionate or valerate fractions in the VFA feed directly translate into higher 3HV content in PHBV, yielding materials that are more flexible and processable compared to PHB. By adjusting the relative proportions of acetate, propionate, and valerate in waste-derived VFA streams, it is therefore possible to tailor the mechanical and thermal performance of PHAs at the metabolic level, without chemical modification of the polymer [13,120].

However, the use of VFAs at high concentrations presents a further challenge, as inhibition primarily arises from the undissociated (protonated) acid fraction rather than the total VFA concentration alone. At pH values close to or below the pK_a of VFAs (~4.7–4.9 for acetic, propionic, and butyric acid), a significant fraction of these acids remains undissociated and can readily diffuse across the cell membrane [111]. Once inside the cytoplasm, which is maintained at a near-neutral pH, the acid dissociates, releasing protons and their corresponding anions. This leads to intracellular acidification, collapse of proton motive force, disruption of enzyme activity, accumulation of anions, and increased ATP demand for active pH homeostasis, thereby inhibiting growth and PHA synthesis. From the literature, growth inhibition is typically observed when the undissociated VFA concentration reaches the low millimolar range (~2–6 mM), although this threshold is strongly strain-dependent [111,121]. Mixed microbial cultures and adapted strains may tolerate higher levels, while sensitive organisms may be inhibited at considerably lower concentrations [113,116]. Notably, odd-chain VFAs, such as propionate and valerate, frequently exert stronger inhibitory effects than acetate at equivalent molar concentrations, due to their higher membrane permeability and intracellular accumulation [111].

Mitigation strategies such as buffering, co-feeding with complementary substrates, or controlled feeding regimes may need to be implemented to improve the suitability of VFA for PHA production [42, 122,123]. A common mitigation strategy is neutralisation of VFAs using bases such as NaOH, to keep the pH levels above the pK_a value of the acid, leading to significant increase in dissociation, reducing the amount of undissociated acid, which can disrupt cellular pH of the microbes, leading to metabolic failure [71]. However, this approach inevitably increases ionic strength and salinity, which can impose additional osmotic stress on microorganisms, impair mass transfer, and complicate downstream processing, including polymer recovery and wastewater management [124]. From a process-engineering perspective, a particularly effective operational strategy is pH-stat fed-batch fermentation, in which VFAs are continuously or semi-continuously supplied only as fast as they are assimilated, while pH is tightly controlled [123,125,126]. This maintains both total VFA concentration and the undissociated fraction below inhibitory thresholds, enabling high volumetric productivities without excessive salt accumulation. In addition to co-feeding with non-acidic carbon sources (e.g., sugars, glycerol) to dilute weak-acid stress, ISPR approaches, such as pertractive extraction

Table 7
Influence of process parameters on growth and properties of accumulated PHAs.

Influencing Process Parameter	Adjustment / Condition	Effect on PHA properties	Research Status	Ref.
VFA composition	Increase in odd-chain VFAs (e.g. propionic acid)	Increases 3HV production, reduces crystallinity and increases toughness.	Requires further research	[110, 111]
Total substrate (VFA) concentration	High loading	Growth inhibition and reduced productivity.	Requires further research	[111]
pH of carbon substrate	pH > 5–6	Lowers undissociated acid toxicity and increases PHA yield.	Well researched	[111]
pH of culture	Neutral to alkaline (strain dependent)	Increases the metabolism of microbes, improving PHA production.	Well researched	[71]
C:N ratio	Nitrogen limitation	Increases intracellular PHA storage.	Well researched	[112]
C:P ratio	Phosphorus limitation	Increases PHA accumulation further.	Well researched	[112]
Dissolved oxygen	Stain specific optimisation	Controls yield and monomer type.	Requires further research	[113]
Temperature	Strain optimum (mesophilic range)	Maximises enzyme activity and growth.	Well researched	[114]
Incubation time	Extended cultivation	Decreases polymer content	Well researched	[115]

or EDBM, offer advanced options for continuously removing VFAs from the broth [127,128]. These techniques simultaneously regulate substrate concentration, reduce toxicity, and improve overall carbon utilisation efficiency.

Additionally, the accumulation and structural properties of PHAs are strongly determined by the nutritional and substrate conditions under which microorganisms are cultivated. Available literature on PHA production suggests that a high carbon-to-nitrogen (C:N) and high carbon-to-phosphorus (C:P) ratio is widely recognised as a key driver of PHA accumulation. Under balanced growth conditions, microbial cultures are typically supplied with a C:N:P ratio of approximately 100:5:1 as it supports the robust PHA formation [112]. During the PHA accumulation phase, nitrogen or phosphorus is deliberately limited while carbon remains in excess, thereby surpassing cell proliferation and redirecting metabolic fluxes toward intracellular carbon storage.

Chen et al. (2019) evaluated PHA production under varying C:N ratios and demonstrated that nitrogen limitation and high C:N ratios enhance PHA accumulation by redirecting carbon flux [129]. Similarly, Ntaikou et al. (2019) confirmed that nitrogen scarcity shifts metabolic pathways toward polymer storage rather than cell proliferation, altering microbial dynamics and increasing production of PHAs, primarily PHB and PHV copolymers [130]. Similarly, this principle applies to phosphorus levels in the feed supplied to the microbes, which is also widely used in PHA production to trigger the storage of biopolymers and is identified as the limiting nutrient that often leads to higher PHA accumulation yields [112,131]. For instance, Kourmentza et al. (2017) investigated the limitation of phosphorus and nitrogen on the yield of PHAs accumulated, demonstrating that nitrogen limitation achieved up to 57% PHA accumulation, while phosphorus limitation achieved up to 70% PHA accumulation in a fed-batch fermentation system [131].

Similarly, oxygen availability is another decisive factor, although its influence is highly strain and process dependent. While aerobic conditions are generally necessary for efficient PHA biosynthesis, the optimal DO level varies markedly between organisms. For instance, for many scl-PHA producers such as *Cupriavidus necator*, insufficient DO leads to reduced cell growth and ultimately lower PHA productivity; thus, adequate aeration is essential to sustain both biomass formation and polymer accumulation. In contrast, several mcl-PHA producers, such as the *Pseudomonas* species, can tolerate or even benefit from moderate oxygen limitation, as reduced DO can shift metabolism toward fatty-acid β-oxidation, influencing monomer composition and favouring mcl-PHA formation. Wijeyekoon et al. (2018) investigated the effect of DO levels on PHA production from HTD or wet air oxidation-derived liquors rich in VFAs [113]. They found that at low DO concentrations, scl-PHA accumulation is induced by reduced adenosine triphosphate (ATP) levels, which limit the anabolic activities required for bacterial growth.

Additionally, incubation time, temperature, and pH are significant factors that may influence PHA accumulation and microbial activity [132]. However, their effect depends on stain selection and microbial physiology. For instance, Mohammed et al. (2020) report that near-neutral conditions (pH ~7) maximised PHA accumulation in *Cupriavidus* and *Bacillus* strains, reflecting the preference of these organisms for neutral environments that support balanced growth and efficient carbon flux towards acetyl-CoA–derived PHA biosynthesis [133]. In contrast, Villano et al. (2010) reported a markedly different response in mixed microbial cultures enriched for *Haloferax.sp* populations, where operation at alkaline conditions (pH 9.5) led to a 48% increase in PHV content [134]. This shift was attributed to pH-driven changes in intracellular redox balance and propionyl-CoA availability, underscoring that pH not only affects microbial growth but also selectively modulates monomer composition depending on the dominant metabolic pathways of the producing strain.

Similarly, Mohanrasu et al. (2020) investigated the effect of fermentation temperature on PHA accumulation, stating that optimal temperatures of ~37 °C show maximum PHA productivity in *Bacillus.sp*, consistent with their mesophilic growth profile and enzyme kinetics [114]. However, lower optimal temperatures (25–30 °C) have been reported for *Pseudomonas putida* and other *Pseudomonas.sp*, where excessive thermal stress can reduce membrane stability and impair mcl-PHA synthesis [135].

Additionally, Odeniyi et al. (2017) demonstrated the significance of incubation time on PHA yield, highlighting that prolonged time may lead to the PHAs accumulated being used as endogenous sources inside the cells [115]. They reported optimal PHA accumulation at 48 h in *Bacillus.sp* culture, with complete depletion observed after 96 h. Comparable trends have been observed in *Cupriavidus necator*, where extended cultivation beyond the stationary phase results in PHA depolymerisation driven by maintenance metabolism [136,137].

3.4. Characterisation of PHAs

PHA yield, accumulation as percentage dry cell weight (%DCW), and monomer composition are widely reported as key process performance indicators, but these metrics are not sufficient on their own to confirm polymer identity or predict material performance. This is particularly important for PHAs produced from waste-derived VFAs, where variations in acid composition, impurities and microbial metabolism can influence monomer incorporation, molecular weight, crystallinity, morphology, and thermal behaviour. Established analytical techniques are routinely applied across the literature to confirm PHA identity, assess structural features, and link polymer composition to thermal and mechanical properties, as illustrated in Fig. 5 [138–140]. Spectroscopic techniques, particularly Fourier-transform infrared spectroscopy (FTIR) and nuclear magnetic resonance (NMR) (^{1}H and ^{13}C), are the primary methods used to confirm PHA chemical identity and monomer composition [138]. FTIR typically shows the characteristic ester carbonyl (C═O) stretching band of PHAs at approximately 1720–1740 cm^{-1}, which is characteristic of aliphatic polyesters, alongside C–O–C stretching vibrations around 1050–1300 cm^{-1} and aliphatic C–H stretching bands around 2850–3000 cm^{-1} [139–141]. However, FTIR

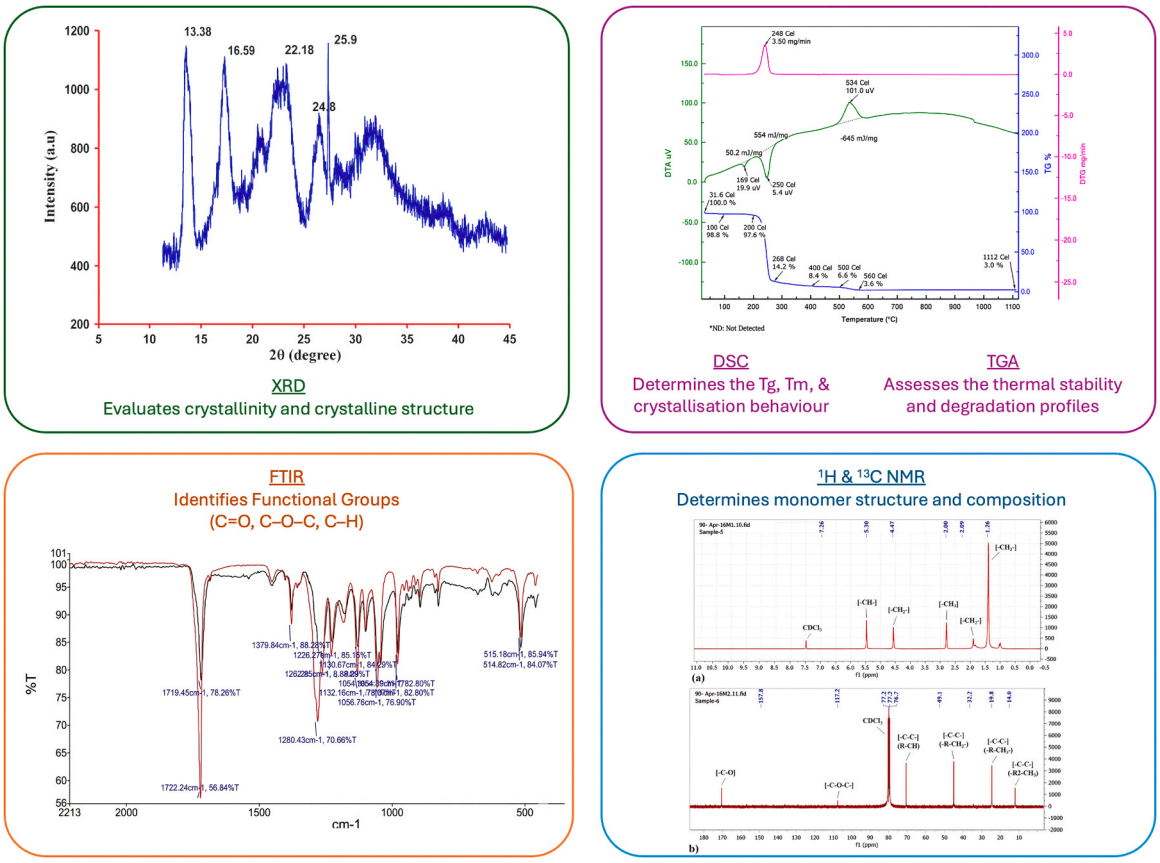


Fig. 5. Representative characterisation techniques used for PHAs, including FTIR, XRD, DSC, TGA, and NMR. Reprinted from [140], *International Journal of Biological Macromolecules*, *Volume* 153 (Copyright 2020) with permission from Elsevier.

alone is not sufficient to fully determine PHA monomer composition or to reliably distinguish between PHB, PHBV, and other PHA copolymers, because many PHAs share similar ester and hydrocarbon functional groups. As a result, FTIR is best used as a rapid screening or confirmatory technique rather than a complete structural identification method. Complementary to FTIR NMR provides more detailed structural confirmation and is particularly valuable for identifying the specific type of PHA produced, because different PHA monomers have distinct side-chain structures that generate distinguishable proton and carbon environments. For PHB, which contains 3HB units with a methyl side group (CH_3), ^{1}H NMR typically shows characteristic signals at approximately 1.2 ppm for methyl protons (CH_3), 2.4–2.6 ppm for methylene protons (CH_2) adjacent to the carbonyl group, and approximately 5.2 ppm for the methine proton (CHO) attached to the ester-bearing carbon [139,140]. In $^{1\ 3}$C NMR, PHB commonly shows carbonyl carbon signals at approximately 169–172 ppm, together with backbone methine, methylene, and methyl carbon signals [139,140]. In PHBV, additional resonances appear because 3HV units contain an ethyl side chain rather than the methyl side chain present in PHB. These additional terminal CH_3 and CH_2 signals from 3HV units allow the 3HB:3HV ratio to be quantified, making NMR particularly useful for distinguishing PHB homopolymer from PHBV copolymer. Medium-chain-length PHAs show further spectral differences because their longer alkyl side chains generate more complex resonances in the alkyl region, particularly around 0.8–1.6 ppm in ^{1}H NMR [142]. These differences arise from the different local chemical environments created by methyl, ethyl, or longer pendant side chains along the polyester backbone linking the carbon source/VFA profile to final polymer structure, where acetate-rich substrates generally favour 3HB formation, propionate and valerate can promote 3HV incorporation, and longer-chain VFAs may contribute to medium-chain-length PHA monomers.

X-ray diffraction (XRD) and scanning electron microscopy (SEM) provide complementary information on crystallinity and morphology [140]. However, XRD does not directly measure mechanical properties such as brittleness or ductility; rather, it provides evidence of crystalline ordering, which strongly influences these properties. Highly crystalline PHB typically exhibits sharp, intense diffraction peaks, reflecting ordered chain packing and restricted polymer chain mobility. This high crystallinity contributes to the stiffness and brittleness commonly associated with PHB. In contrast, incorporation of 3HV units into PHBV disrupts the regular 3HB chain packing, leading to broader, less intense XRD peaks and a lower degree of crystallinity. This reduction in crystallinity increases chain mobility and is commonly associated with greater flexibility and ductility than in PHB. This effect is routinely confirmed by broader, less intense diffraction peaks in XRD patterns, as observed by Maity et al., 2020 [140]. Analysis such as SEM can reveal polymer morphology, surface roughness, porosity, and phase separation in blends or composites. However, SEM does not confirm chemical identity and should not be used alone to prove PHA formation. Instead, it is most useful as a supporting technique for assessing the physical appearance and morphology of the extracted polymer, particularly when waste-derived substrates may introduce impurities, heterogeneous polymer fractions, or extraction residues.

Thermal characterisation by differential scanning calorimetry (DSC) and thermogravimetric analysis (TGA) inks polymer structure to processing behaviour and end-use suitability. Data from DSC analysis provides glass transition temperature (Tg), melting temperature (Tm), and crystallisation behaviour, all of which are strongly influenced by monomer composition and crystallinity [140,143]. For instance, PHB typically exhibits a Tm of ~170–180 °C and a Tg near −5 to + 5 °C [144]. Increasing incorporation of 3HV units progressively reduces both thermal transitions, with Tm decreasing to ~120–160 °C and Tg shifting

to ~−10 to −20 °C at higher 3HV fractions (~30–50 mol%) [143,145], reflecting reduced crystallinity and increased chain mobility. Similarly, TGA provides insight into thermal stability and degradation behaviour, which are critical for processing and end-use applications. The literature suggests that commonly reported TGA values for PHB-based polymers typically range from ~150 to ~350 °C, with maximum degradation occurring at ~467 °C [143,146]. These shifts are significant because they indicate reduced crystallinity, increased chain mobility, and potentially improved flexibility compared with brittle PHB. Biopolymers such as Mcl-PHAs generally exhibit lower melting temperatures and greater elastomeric behaviour than scl-PHAs, further demonstrating how monomer structure determines final material properties [142,147]. This is particularly important for waste-derived VFA systems, where residual organics, salts, pigments, inhibitory compounds, or chain-terminating impurities may reduce polymer purity, alter degradation behaviour, or broaden molecular weight distributions.

In addition to spectroscopic, morphological, crystalline, and thermal analyses, molecular weight characterisation is important for assessing the quality and processability of recovered PHAs. Gel permeation chromatography (GPC), a type of size-exclusion chromatography (SEC), is commonly used for polymer characterisation, including molecular weight determination and polydispersity (number-average/weight-average) of PHAs, which influence melt strength, toughness, degradation behaviour, and processing stability [148,149]. Reported PHA molecular weights (~50–3000 kDa), polydispersity index (1.5–3.0), and number-average molecular weights (50–1000 kg/mol) vary significantly, depending on the microbial strain, substrate, cultivation conditions, extraction method, and polymer composition [148]. Biopolymers such as PHB and PHBV typically have higher molecular weights, contributing to their high crystallinity and brittleness, compared to mcl-PHAs (~50–100 kDa) [142]. For waste-derived VFA systems, GPC/SEC is particularly relevant because impurities, nutrient limitations, inhibitory compounds, or chain-transfer reactions may reduce polymer chain length or broaden the molecular weight distribution. As a result, GPC/SEC can help determine whether PHAs produced from waste-derived VFAs are comparable in quality to polymers produced from pure substrates.

Collectively, these characterisation techniques establish a direct link between feedstock composition, microbial conversion pathways, and final material properties. In the context of waste-derived VFAs, this link is particularly important, as variations in acid composition, such as acetate versus propionate or valerate, influence monomer incorporation and therefore determine whether the product is mainly PHB, PHBV, or another PHA type. At the same time, impurities in waste-derived streams may affect microbial metabolism, polymer accumulation, molecular weight, crystallinity, and thermal stability. At minimum, FTIR and NMR are recommended in conjunction for chemical confirmation and monomer identification; DSC and XRD are recommended for assessing crystallinity and thermal behaviour; TGA is recommended for effective evaluation of thermal stability; and, where possible, GPC/SEC is recommended for determining the molecular weight and molecular weight distribution.

3.5. Potential applications across sectors

Representing a promising class of biodegradable and biocompatible polyesters, PHAs have diverse applications across several industrial sectors. A distinct advantage of PHAs produced from VFA-derived feedstocks is the ability to tailor their monomer composition by adjusting fermentation conditions and substrate ratios. This tunability allows the optimisation of mechanical and thermal properties for specific end uses, where higher 3HV monomer content leads to reduced crystallinity, improving flexibility and making the polymer more suited to soft film applications [150]. Alternatively, research on the monomer composition of PHAs links the relationship between higher content of 3HB polymers and the resulting crystalline, more rigid structures, making it unsuitable for certain applications requiring flexibility [150]. However, the use of waste-derived VFAs as carbon substrates for PHA production can introduce residual impurities, such as odorous compounds or pigments, which may influence the colour, smell, or consistency of the resulting polymers [151]. Consistency and purity of PHAs are especially crucial within the biomedical field, as stringent requirements for biomedical-grade PHAs highlight that waste-derived feedstocks require additional purification and monomer control to meet medical standards [10,152]. Currently, most established applications of PHAs are in the packaging sector, where they are used for films, coatings, disposable cutlery, straws, and food containers. This sector has seen significant commercial investment, with companies such as Danimer Scientific (USA) [153,154], producer of Nodax PHA, and CJ Biomaterials (South Korea) [155], producer of PHACT, who are actively supplying materials for consumer packaging and food-contact products. Other notable players include RWDC Industries (US) [156], producing PHAs for straws and films, and Kaneka Corporation (Japan) [157], which manufactures PHAs for packaging and agricultural films. These companies have formed strategic partnerships with global consumer brands, underlining the growing role of PHAs in replacing single-use plastics.

Another significant area of PHA application is in the biomedical and pharmaceutical industries, where their inherent biocompatibility and adjustable degradation profiles make them attractive for resorbable sutures, scaffolds, and drug delivery systems [158]. Tepha, Inc. (US) has pioneered the commercialisation of medical-grade PHAs for surgical meshes, sutures, and scaffolds, with several products already approved for medical use and clinically deployed [159]. Similarly, biotechnology firms such as Bluepha (China) are developing marine-degradable PHA polymers that may have future applications in biomedical and consumer health products, given that they are exceptionally pure and compositionally consistent with relative medical regulations [160]. The integration of PHAs into medical devices underscores their added value in high-performance applications, surpassing that of commodity plastics.

Additionally, PHAs are finding applications in agriculture and environmental sectors, where their soil-biodegradability is harnessed for mulch films, controlled-release fertilisers, and biodegradable plant pots [160]. European initiatives, such as the COM4PHA project, coordinated by organisations including Venvirotech and Aimplas, are developing PHBV-based films and coatings for use in agricultural contexts [161,162]. These waste-to-PHA pathways align with circular economy principles and are particularly attractive in the context of climate and waste management policies. While research continues into PHA composites for high-value applications, including functional coatings, sensors, and additive manufacturing filaments, large-scale commercialisation within these sectors is still emerging [158,160].

4. Waste-derived VFAs vs. alternative carbon sources

Despite the strong potential of waste-derived VFAs as sustainable carbon substrates, the industrialisation of VFA-based PHA production remains constrained by several interconnected bottlenecks. These challenges arise primarily from feedstock variability and impurity load, process instability and microbial inhibition, and downstream processing and environmental trade-offs. Unlike conventional substrates, these limitations are intrinsic to the waste-to-VFA pathway and must be addressed to enable scalable and economically viable PHA production. This section critically evaluates these barriers from both technical, environmental, and economic perspectives (summarised in Fig. 6).

4.1. Process efficiency and technical performance

Under controlled conditions, VFAs can match or exceed other carbon sources, such as sugar and oil, for PHA production in terms of %DCW and characteristic tunability. However, results are highly dependent on system parameters, including culture, pH, DO, feeding, and inhibitor

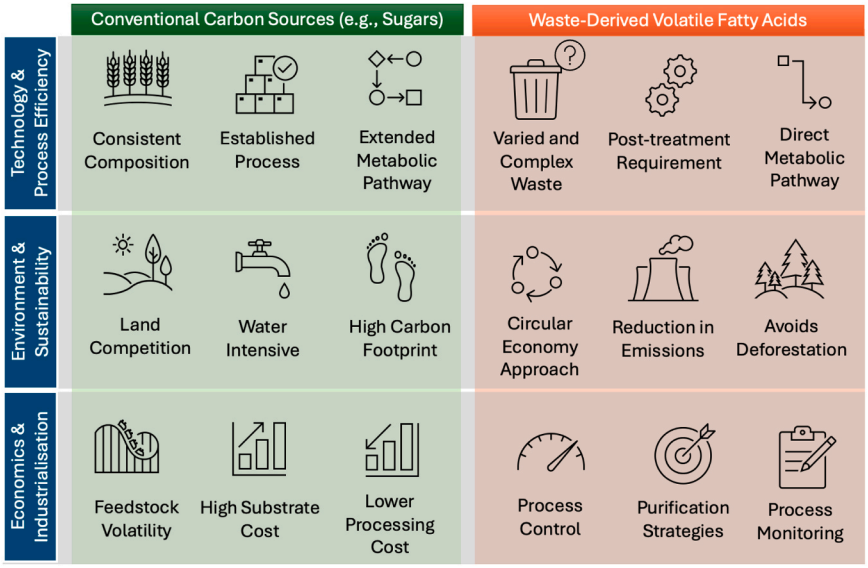


Fig. 6. Comparison of conventional carbon sources for PHA production to waste-derived VFAs.

load [163,164]. For instance, studies suggest that VFAs, such as acetic, butyric, and propionic acids, are more closely related metabolically to PHA building blocks than sugars like glucose, shortening the metabolic pathway and leading to higher substrate-to-PHA conversion efficiencies [165,166]. Studies report up to 89% PHA accumulation using tailored VFA mixtures and carefully selected bacterial strains [104,105]. Alternatively, PHAs from glucose or oils often reach 70–80% accumulation in pure cultures, with more consistent growth kinetics and shorter lag phases, allowing for easier control of culture conditions and optimisation of yields [167,168].

However, while these alternative carbon sources confer many advantageous PHA production traits, they also have limitations in terms of PHA accumulation and production. For instance, lignocellulosic hydrolysates commonly contain inhibitory compounds such as hydroxymethylfurfural (HMF), furans, and phenolics that require detoxification prior to microbial exposure, as they may inhibit growth and PHA accumulation [169–171]. Additionally, substrates such as crude glycerol can be contaminated with methanol, soaps, and salts, which can affect microbial performance and downstream processing [172]. In contrast, unmodified plant oils tend to prefer production of mcl-PHA unless the microbial strain is specifically engineered or selected [173]. These feedstock-specific impurities and metabolic biases can reduce effective yields, necessitate additional pre-treatment steps, and influence process economics despite the generally steadier growth kinetics of sugar- and oil-based systems. Similarly, under less optimised conditions, the complexity, toxicity, and inconsistency of waste-derived VFAs may lead to lower microbial growth rates and, consequently, lower PHA accumulation yields [163]. Waste streams often contain ammonia, heavy metals, sulphides, phenolic compounds, and long-chain fatty acids, which can inhibit the microbial fermentation processes used to produce PHAs from VFAs. These compounds can damage cell membranes, interfere with enzymatic activity, and disrupt metabolic pathways [163]. This sensitivity to impurities and undissociated acids represents a major scale-up barrier, as maintaining stable microbial performance requires continuous control of feed composition, pH, and inhibitor levels, conditions that are difficult to guarantee with heterogeneous waste streams. For instance, Agustín Martinez et al. (2015) reported that polyphenols present in olive mill wastewater inhibited PHA accumulation in cells by up to 30% of DCW [174]. To ensure VFAs are suitable for microbial PHA synthesis, a pretreatment and a carefully selected VFA recovery technique, discussed in this review, such as EDBM or pertraction, is often required to recover acids, remove solids, suspended particles, and inhibitory compounds. Pretreatment techniques include pH-stat fed batch and neutralisation to limit undissociated acid toxicity, selective adsorption of inhibitory compounds, fractionation, and nutrient conditioning, which can sustainably enhance PHA yields. Additionally, mitigation strategies could include selecting robust microbial systems, especially engineered strains with enhanced improved tolerance to inhibitors. For instance, Chen et al. (2018) explored the use of engineered E. coli for the production of PHB and PHBV using acetate as a main carbon source [175]. The research identified that the engineered strain could co-metabolise substrates such as acetate, propionate, and butyrate in untreated mixtures, producing copolymers such as PHB-co-PHV with adjustable composition. The primary technical bottleneck, therefore, lies in the trade-off between metabolic efficiency and process robustness. While VFAs enable high theoretical conversion efficiencies, their variability, impurity content, and inhibitory effects significantly reduce process stability under industrial conditions.

4.2. *Environmental and sustainability considerations*

The environmental impacts of PHAs are closely linked to their end-of-life behaviour, which distinguishes them from conventional petrochemical plastics. As bio-based and biodegradable polyesters, PHAs can be metabolised by a wide range of microorganisms, enabling their conversion into carbon dioxide, water, and biomass under aerobic conditions, or into methane under anaerobic conditions [176]. This biodegradability provides a clear sustainability advantage by reducing long-term plastic persistence and microplastic accumulation in the environment. However, it is critical to note that PHA biodegradation is highly dependent on the environmental conditions [176,177]. Effective degradation typically occurs under high microbial activity, elevated temperatures (in the thermophilic range), high humidity, and aerobic conditions [176]. However, certain PHA grades have also demonstrated biodegradability in soil, freshwater, and marine environments [177]. Consequently, PHAs can support circular end-of-life pathways, particularly when integrated into managed organic waste streams such as industrial composting or anaerobic digestion, where both material recovery and energy recovery are possible [178].

Beyond end-of-life considerations, the environmental impacts and overall sustainability of PHAs are also strongly influenced by the choice of carbon feedstock used during their production, as different feedstocks, such as waste-derived VFAs versus conventional substrates (such

as sugars or plant oils) for PHA production, entail distinct resource demands, energy inputs, and waste valorisation potential. Waste-derived VFAs represent a secondary resource, meaning their use does not compete with food systems and does not require dedicated cultivation. Conversely, alternative carbon sources, like glucose, sucrose, glycerol, and plant oils, are typically derived from primary agricultural production. The cultivation of these crops requires significant resource inputs, consuming approximately 1000–3000 L of water per kilogram of sugar and emitting 1.2–1.8 kg CO_2-eq per kilogram of glucose produced [179]. Life cycle assessment studies consistently demonstrate that replacing crop-derived substrates with waste-derived VFAs can reduce total GHG emissions by 30–60% and lower cumulative energy demand by up to 50% in PHA production [25,180].

Additionally, transforming organic waste into PHA feedstock, such as VFAs, closes the material loop and contributes significantly to the implementation of a circular economy, in which materials are valorised as a functional resource rather than discarded. By recovering carbon and nutrients from organic residues, VFA-based systems close material loops, transforming low-value waste streams into high-value, biodegradable polymers that can be reintroduced into waste streams or treatment facilities at the end of their life [181]. This integration promotes waste minimisation, resource efficiency, and reduced dependency on virgin feedstocks, aligning with regulations and policies set by environmental organisations that promote responsible consumption and production of materials. For instance, the EU Circular Economy Action Plan 2020 promotes keeping resources in use and minimising waste by extending product life cycles and recycling materials rather than landfilling or incinerating them [182]. Converting organic waste into VFAs, which are used as feedstock for PHA production, valorises waste as a resource rather than discarding it, directly supporting closed-loop resource use [183]. Additionally, Sustainable Development Goal 12 (SDG 12) encourages efficient use of natural resources, turning waste into valuable feedstock and reducing raw resource extraction, which is directly supported by the waste-derived VFA-to-PHA chain [184].

Conversely, conventional carbon sources such as glucose or plant oils, though renewable, remain embedded in linear production systems characterised by intensive monoculture cultivation, centralised processing, and fossil fuel–dependent logistics. Their limited capacity for material recovery restricts their contribution to circular economy objectives, unless derived from industrial by-products such as crude glycerol or lignocellulosic residues [181]. Additionally, extracting VFAs from waste streams helps avoid deforestation, habitat loss, and soil degradation, whereas alternative carbon sources often lead to land expansion. Sugarcane and palm oil cultivation are linked to deforestation in tropical regions, threatening biodiversity and carbon sinks. Even when grown on marginal lands, these crops can displace native vegetation or compete with food production [181,185]. However, a key environmental bottleneck emerges when considering the full process chain. The need for VFA recovery, purification, and conditioning can introduce significant energy demand, chemical usage, and secondary waste streams, partially offsetting the environmental benefits of waste valorisation. At an industrial scale, these additional processing requirements complicate system integration and may reduce the overall sustainability advantage compared to more conventional, compositionally consistent substrates.

4.3. Economic viability and industrialisation barriers

The economic feasibility of PHA production from waste-derived VFAs presents a compelling alternative to conventional feedstocks, particularly when implemented into circular economy models. Waste-derived VFAs are often associated with low-cost substrates, as they are generated from readily available waste streams that may be subject to gate-fee incentives, under which waste generators pay for disposal. This significantly reduces raw material costs, which account for $>$ 50% of the total PHA production expenses [186,187]. In contrast, conventional carbon feedstocks offer more predictable composition and fermentation performance, resulting in higher, more consistent PHA yields and simpler downstream processing [181]. However, their reliance on agricultural or refined carbon sources introduces volatility linked to food commodity prices and land use impacts, introducing high initial costs. Ozturk et al. (2025) indicate that while sugar fermentation is the most established commercial route, currently dominating ~50% of the current market, production costs rarely fall below $4.00 USD/kg without significant government subsidies [188]. Alternatively, although plant oils are more expensive per unit mass than sugars, they are significantly more energy-dense. Microbes can convert oils to PHA with much higher yields (sometimes exceeding 0.6 – 0.8 g PHA / g oil) as demonstrated in Table 8. This higher productivity enables higher cell densities in the bioreactor, which can lower per-unit production costs compared to sugars. Research suggests that vegetable oil fermentation is a preferred route for mcl-PHAs, which command premium pricing in biomedical and specialty packaging markets, as shown in Table 8 [189,190]. In a circular economy model, the cost of VFAs as a feedstock is negligible or even negative, as producers are often paid to process this waste. However, waste-derived VFAs, while cheaper, are economically sensitive to compositional variability caused by differences in waste source and treatment parameters. However, these cost savings are offset by challenges associated with VFA composition and variability, which require greater process control, pretreatment, recovery from waste-streams using expensive technologies, and monitoring, depending on the feedstock type and treatment conditions [181]. Under optimised PHA production conditions, costs range from $1.20 to $2.30 USD/kg. However, for high-purity applications, the cost rises toward $4.50 USD/kg due to complex purification and downstream processing needs. Studies report that pilot-scale systems using waste-derived VFAs have successfully demonstrated costs under $2.50 USD/kg, making them the most competitive alternative to conventional plastics like PP [163].

This variability represents a primary technical and economic bottleneck in PHA industrialisation, since the transition from batch to continuous or large-scale feeding regimes is often hindered by the unpredictable kinetics of waste-derived substrates. Specifically, fluctuations in VFA composition demand enhanced regulation of parameters such as pH and DO during fermentation to maintain microbial performance. These additional control measures increase both capital expenditure, due to investment in advanced monitoring and control systems, and operating expenditure, as they consume more energy and reagents. Furthermore, ensuring consistent polymer quality and optimising separation, filtration, and cleanup are often necessary to remove impurities. The downstream processing stage constitutes a major economic bottleneck, as the presence of complex organic and inorganic contaminants in waste streams necessitates aggressive purification protocols to either remove harmful additives or isolate VFAs. This additional purification also raises capital costs through upgraded equipment and operational expenditures through higher maintenance and consumable costs [181]. The high energy intensity of these recovery steps negates the

Table 8
Economic comparison of PHA production from various carbon sources.

Carbon Source	Estimated Feedstock Cost (USD/kg)	Estimated PHA Production Cost (USD/kg)	Economic Bottleneck	Ref.
Refined sugars (glucose/sucrose)	0.4–0.6	10–15	High raw material cost and competition with food supply.	[188]
Conventional plant oils	0.6–0.9	12–18	Feedstock price volatility.	[189,190]
Waste-derived VFAs	$<$ 0.10 (or negative *)	5–10	High downstream processing and purification costs.	[163]

* Negative cost refers to gate fees received for waste treatment.

carbon-footprint advantages of utilising waste-derived feedstocks for PHA production.

Substrates such as sugars and oils enable predictable fermentation with high PHA content in cells, which can lower bioprocessing and purification costs, but at the expense of higher feedstock prices and potentially increased environmental impacts. Moreover, PHAs derived from waste-based VFAs may encounter stricter regulatory scrutiny for applications involving food contact or biomedical use, as potential contaminants from waste streams raise safety and quality concerns [95]. This scepticism is heightened by the lack of established certification standards or quality assurance protocols specific to waste-derived feedstocks. Implementing clear benchmarks for product quality and safety may encourage manufacturers and end-users to integrate such materials into their production lines, thereby limiting potential performance variability, contamination risks, and reputational concerns surrounding PHA products synthesised via waste-derived VFAs.

5. Future perspectives and research directions

To transition utilisation of waste-derived VFAs for PHA production from a promising laboratory concept to a staple of the global bioeconomy, research should shift toward integrated, innovative strategies that address the systemic complexities of the process. Due to the primary barrier identified, the compositional variability of waste feedstocks, future research should focus on developing predictive models that can adjust fermentation parameters (e.g., pH-stat feeding or dissolved oxygen levels) in real-time. By utilising sensor data to predict the resulting polymer's monomeric profile, researchers can ensure material consistency, a prerequisite for high-value applications in the medical and food-packaging sectors where purity and mechanical standards are necessary [191,192]. Furthermore, future advancements in PHA production may move toward engineered microbial consortia and synthetic biology. While current studies often utilise pure cultures or natural mixed cultures, there is significant potential in engineering robust strains specifically designed to tolerate the inhibitory compounds (e.g., ammonia and heavy metals) typically found in waste-derived liquors [193]. From an industrial standpoint, the future of this technology depends on integrating PHA synthesis into multi-product biorefineries. Rather than viewing PHA as a standalone product, integrating its production with other waste-valorisation streams, such as bio-hydrogen recovery or the conversion of residual fermentation solids into biogas, could significantly improve the facility's techno-economic outlook [194,195]. Such a holistic approach would help bridge the current price gap between bioplastics and petroleum-based alternatives, which remains a critical commercial hurdle towards industrialisation.

6. Conclusion

This review summarises evidence for the potential of utilising waste-derived volatile fatty acids (VFAs) as a sustainable and economically viable feedstock for the biosynthesis of polyhydroxyalkanoate (PHA) bioplastics. The strategy aligns robustly with circular economy principles by offering a pathway to support lower-impact PHA production from wastes, reducing reliance on virgin, conventional, and food-competing carbon sources. Primarily generated through biological and thermochemical treatment of diverse waste streams, VFAs are direct metabolic precursors for PHA production, enabling high substrate-to-PHA conversion efficiencies that can rival or exceed those of traditional sugar-based substrates under optimised conditions. While anaerobic digestion is the most conventional method for VFA production from biological waste streams, thermochemical treatment methods such as HTD show significant promise for converting more complex waste streams into VFAs. Crucially, the VFA profile can be manipulated to produce tailored PHA copolymers (e.g., PHBV and PHB). The use of propionate and valerate substrates increases the 3HV content in PHBV biopolymers, thereby reducing crystallinity and increasing toughness. The utilisation of acetate substrates results in higher PHB polymer production, increased stiffness, and a higher tensile strength. Utilisation of VFAs offers adjustable mechanical properties (flexibility, ductility) suitable for varied applications, including packaging, agriculture, and biomedical devices. The adoption of waste-derived VFAs offers compelling environmental benefits, including waste valorisation, a reduced carbon footprint, and an attractive cost advantage over conventional carbon sources available for PHA production. Despite the clear promise, the transition to commercial-scale PHA production using waste-derived VFAs is hindered by persistent technical and economic challenges. Separation techniques focused on selective VFAs, such as EDBM and pertraction, can retain undissociated acids in the culture medium at low concentrations suitable for microbial synthesis. Additionally, microbial culture modification strategies, such as pH-stat fed-batch for toxicity control, can be implemented to increase intracellular metabolism, as well as monomer control via selective VFA dosing (e.g., propionate and valerate) to achieve targeted PHA structures. Additional evaluation strategies, such as LCA and techno-economic assessment (TEA), are recommended for further successful development and implementation of waste-derived VFA-to-PHA production.

CRediT authorship contribution statement

Milica Mihajlovic: Writing – original draft, Methodology, Investigation, Formal analysis. **Kaveh Shahbaz:** Writing – review & editing, Supervision. **Céline Vaneeckhaute:** Writing – review & editing, Supervision. **Saeid Baroutian:** Writing – review & editing, Supervision, Conceptualization.

Declaration of Competing Interest

The authors declare that they have no known competing financial interests or personal relationships that could have appeared to influence the work reported in this paper.

I have nothing to declare.

Acknowledgements

The authors would like to acknowledge the financial support of Dick and Mary Earle and express sincere gratitude towards the Dick and Mary Earle Scholarship committee.

Data availability

No data was used for the research described in the article.

References

[1] UN Environement Programme, Taking on plastic pollution | UNEP Annual Report, (2024). ⟨https://www.unep.org/annualreport/2024/stories/taking-plastic-pollution⟩ (accessed October 20, 2025).
[2] Kitech, In 2024, The Global Plastic Production Will Exceed 400 Million Tons, (2025). ⟨https://www.kitechrecycle.com/news/in-2024-the-global-plastic-production-will-ex-84661069.html⟩ (accessed October 20, 2025).
[3] European Environment Agency, Plastics, (2025). ⟨https://www.eea.europa.eu/en/circularity/sectoral-modules/plastics⟩ (accessed October 20, 2025).
[4] UN Environment, Plastic Pollution, Plast. Pollut. (2022). ⟨https://www.unep.org/plastic-pollution⟩ (accessed October 20, 2025).
[5] Ministry for the Environment, Towards an international treaty on plastic pollution, Minist. Environ. (2025). ⟨https://environment.govt.nz/what-government-is-doing/international-action/towards-an-international-treaty-on-plastic-pollution/⟩ (accessed October 20, 2025).
[6] C. Du, Z. Wang, X. Zhuo, R. Geng, C. Liu, H. Wang, D. Chu, S. Li, R. Li, G. Zheng, D. Ding, Biosynthesis of polyhydroxyalkanoates (PHAs) from organic waste-derived volatile fatty acids (VFAs), Green. Chem. 27 (2025) 1939–1968, https://doi.org/10.1039/D4GC05125D.
[7] A. Anjum, M. Zuber, K.M. Zia, A. Noreen, M.N. Anjum, S. Tabasum, Microbial production of polyhydroxyalkanoates (PHAs) and its copolymers: A review of recent advancements, Int. J. Biol. Macromol. 89 (2016) 161–174, https://doi.org/10.1016/j.ijbiomac.2016.04.069.

[8] Z.A. Raza, S. Abid, I.M. Banat, Polyhydroxyalkanoates: Characteristics, production, recent developments and applications, Int. Biodeterior. Biodegrad 126 (2018) 45–56, https://doi.org/10.1016/j.ibiod.2017.10.001.
[9] L. Vogli, S. Macrelli, D. Marazza, P. Galletti, C. Torri, C. Samorì, S. Righi, Life cycle assessment and energy balance of a novel polyhydroxyalkanoates production process with mixed microbial cultures fed on pyrolytic products of wastewater treatment sludge, Energies 13 (2020) 2706, https://doi.org/10.3390/en13112706.
[10] S. Gautam, A. Gautam, J. Pawaday, R.K. Kanzariya, Z. Yao, Current status and challenges in the commercial production of polyhydroxyalkanoate-based bioplastic: a review, Processes 12 (2024) 1720, https://doi.org/10.3390/pr12081720.
[11] A.A. Moronkola, K.F.K. Oyedeko, R.J. Patinvoh, J.G. Akinbomi, S. Wainaina, M. J. Taherzadeh, Optimization of volatile fatty acids from food wastes for polyhydroxyalkanoates synthesis, Biomass-.-. Convers. Biorefinery 15 (2025) 20247–20264, https://doi.org/10.1007/s13399-025-06548-y.
[12] B. Chavez, V. Raghavan, B. Tartakovsky, A comparative analysis of biopolymer production by microbial and bioelectrochemical technologies, RSC Adv. 12 (2022) 16105–16118, https://doi.org/10.1039/d1ra08796g.
[13] K. Szacherska, P. Oleskowicz-Popiel, S. Ciesielski, J. Mozejko-Ciesielska, Volatile Fatty Acids as Carbon Sources for Polyhydroxyalkanoates Production, Polymers 13 (2021) 321, https://doi.org/10.3390/polym13030321.
[14] Sandra Lepak-Kuc, Katarzyna Wójkowska, Dorota Biernacka, Aleksandra Kadziela, Tadeusz Murawski, Daniel Janczak, Ma. łgorzata Jakubowska, Biodegradable Conductive Layers Based on a Biopolymer Polyhydroxybutyrate/Polyhydroxyvalerate and Graphene Nanoplatelets Deposited by Spray-Coating Technique, ResearchGate, 2025, https://doi.org/10.3390/coatings13101791.
[15] A.L. Rivera-Briso, Á. Serrano-Aroca, Poly(3-Hydroxybutyrate-co-3-Hydroxyvalerate): Enhancement Strategies for Advanced Applications, Polymers 10 (2018) 732, https://doi.org/10.3390/polym10070732.
[16] B. Andreeßen, N. Taylor, A. Steinbüchel, Poly(3-Hydroxypropionate): a Promising Alternative to Fossil Fuel-Based Materials, Appl. Environ. Microbiol 80 (2014) 6574–6582, https://doi.org/10.1128/AEM.02361-14.
[17] A.G. Aduhene, H. Cui, H. Yang, C. Liu, G. Sui, C. Liu, Poly(3-hydroxypropionate): Biosynthesis Pathways and Malonyl-CoA Biosensor Material Properties, Front. Bioeng. Biotechnol. 9 (2021), https://doi.org/10.3389/fbioe.2021.646995.
[18] S. Saito, R. Imai, Y. Miyahara, M. Nakagawa, I. Orita, T. Tsuge, T. Fukui, Biosynthesis of Poly(3-hydroxybutyrate-co-3-hydroxyhexanoate) From Glucose by Escherichia coli Through Butyryl-CoA Formation Driven by Ccr-Emd Combination, Front. Bioeng. Biotechnol. 10 (2022), https://doi.org/10.3389/fbioe.2022.888973.
[19] H.J. Tang, S.Z. Neoh, K. Sudesh, A review on poly(3-hydroxybutyrate-co-3-hydroxyhexanoate) [P(3HB-co-3HHx)] and genetic modifications that affect its production, Front. Bioeng. Biotechnol. 10 (2022), https://doi.org/10.3389/fbioe.2022.1057067.
[20] R. Rai, D.M. Yunos, A.R. Boccaccini, J.C. Knowles, I.A. Barker, S.M. Howdle, G. D. Tredwell, T. Keshavarz, I. Roy, Poly-3-hydroxyoctanoate P(3HO), a Medium Chain Length Polyhydroxyalkanoate Homopolymer from Pseudomonas mendocina, Biomacromolecules 12 (2011) 2126–2136, https://doi.org/10.1021/bm2001999.
[21] A. Bagdadi, M. Safari, P. Dubey, P. Basnett, P. Sofokleous, E. Humphrey, I. Locke, M. Edirisinghe, C. Terracciano, A. Boccaccini, J. Knowles, S. Harding, I. Roy, Poly (3-hydroxyoctanoate), a promising new material for cardiac tissue engineering, J. Tissue Eng. Regen. Med. 12 (2016), https://doi.org/10.1002/term.2318.
[22] R.P. Mahato, S. Kumar, P. Singh, Production of polyhydroxyalkanoates from renewable resources: a review on prospects, challenges and applications, Arch. Microbiol 205 (2023) 172, https://doi.org/10.1007/s00203-023-03499-8.
[23] C. Maraveas, Environmental Sustainability of Greenhouse Covering Materials, Sustainability 11 (2019) 6129, https://doi.org/10.3390/su11216129.
[24] A. Vázquez-Fernández, M.E. Suárez-Ojeda, J. Carrera, Review about bioproduction of Volatile Fatty Acids from wastes and wastewaters: Influence of operating conditions and organic composition of the substrate, J. Environ. Chem. Eng. 10 (2022) 107917, https://doi.org/10.1016/j.jece.2022.107917.
[25] W.S. Lee, A.S.M. Chua, H.K. Yeoh, G.C. Ngoh, A review of the production and applications of waste-derived volatile fatty acids, Chem. Eng. J. 235 (2014) 83–99, https://doi.org/10.1016/j.cej.2013.09.002.
[26] I. Owusu-Agyeman, E. Plaza, Z. Cetecioglu, Production of volatile fatty acids through co-digestion of sewage sludge and external organic waste: Effect of substrate proportions and long-term operation, Waste Manag 112 (2020) 30–39, https://doi.org/10.1016/j.wasman.2020.05.027.
[27] S.K. Bhatia, Y.-H. Yang, Microbial production of volatile fatty acids: current status and future perspectives, Rev. Environ. Sci. Biotechnol. 16 (2017) 327–345, https://doi.org/10.1007/s11157-017-9431-4.
[28] G. Bravo-Porras, L.A. Fernández-Güelfo, C.J. Álvarez-Gallego, M. Carbú, D. Sales, L.I. Romero-García, Influence of the total concentration and the profile of volatile fatty acids on polyhydroxyalkanoates (PHA) production by mixed microbial cultures, Biomass Convers. Biorefinery 14 (2024) 239–253, https://doi.org/10.1007/s13399-021-02208-z.
[29] V. Ashokkumar, G. Flora, R. Venkatkarthick, K. SenthilKannan, C. Kuppam, G. Mary Stephy, H. Kamyab, W.-H. Chen, J. Thomas, C. Ngamcharussrivichai, Advanced technologies on the sustainable approaches for conversion of organic waste to valuable bioproducts: Emerging circular bioeconomy perspective, Fuel 324 (2022) 124313, https://doi.org/10.1016/j.fuel.2022.124313.
[30] G. Kumar, V.K. Ponnusamy, R.R. Bhosale, S. Shobana, J.-J. Yoon, S.K. Bhatia, J. Rajesh Banu, S.-H. Kim, A review on the conversion of volatile fatty acids to polyhydroxyalkanoates using dark fermentative effluents from hydrogen production, Bioresour. Technol. 287 (2019) 121427, https://doi.org/10.1016/j.biortech.2019.121427.
[31] J. Wang, S. Liu, J. Huang, Z. Qu, A review on polyhydroxyalkanoate production from agricultural waste Biomass: Development, Advances, circular Approach, and challenges, Bioresour. Technol. 342 (2021) 126008, https://doi.org/10.1016/j.biortech.2021.126008.
[32] S.K. Bhatia, S.V. Otari, J.-M. Jeon, R. Gurav, Y.-K. Choi, R.K. Bhatia, A. Pugazhendhi, V. Kumar, J. Rajesh Banu, J.-J. Yoon, K.-Y. Choi, Y.-H. Yang, Biowaste-to-bioplastic (polyhydroxyalkanoates): Conversion technologies, strategies, challenges, and perspective, Bioresour. Technol. 326 (2021) 124733, https://doi.org/10.1016/j.biortech.2021.124733.
[33] G.K. Veluswamy, K. Shah, A.S. Ball, A.J. Guwy, R.M. Dinsdale, A techno-economic case for volatile fatty acid production for increased sustainability in the wastewater treatment industry, Environ. Sci. Water Res. Technol. 7 (2021) 927–941, https://doi.org/10.1039/D0EW00853B.
[34] S. Agnihotri, D.-M. Yin, A. Mahboubi, T. Sapmaz, S. Varjani, W. Qiao, D.Y. Koseoglu-Imer, M.J. Taherzadeh, A glimpse of the world of volatile fatty acids production and application: a review, Bioengineered 13 (n.d.) 1249–1275. https://doi.org/10.1080/21655979.2021.1996044.
[35] A.C. Dimian, A.A. Kiss, Enhancing the separation efficiency in acetic acid manufacturing by methanol carbonylation, Chem. Eng. Technol. 44 (2021) 1792–1802, https://doi.org/10.1002/ceat.202100230.
[36] Mordor Intelligence, Acetic acid market size, growth, share & trends report 2031, Mordor Intell. (2026). ⟨https://www.mordorintelligence.com/industry-reports/acetic-acid-market⟩ (accessed January 29, 2026).
[37] S. U.-D, W. L, D. Gn, Volatile Fatty Acid Production from Anaerobic Digestion of Organic Residues (Clifton NJ), Methods Mol. Biol. 1995 (2019), https://doi.org/10.1007/978-1-4939-9484-7_20.
[38] N.-J. Kim, S.-J. Lim, H.N. Chang, Volatile Fatty Acid Platform: Concept and Application. Emerg. Areas Bioeng, John Wiley & Sons, Ltd, 2018, pp. 173–190, https://doi.org/10.1002/9783527803293.ch10.
[39] Q. Zhang, Y. Lu, X. Zhou, X. Wang, J. Zhu, Effect of different vegetable wastes on the performance of volatile fatty acids production by anaerobic fermentation, Sci. Total Environ. 748 (2020) 142390, https://doi.org/10.1016/j.scitotenv.2020.142390.
[40] S. Greses, E. Tomás-Pejó, C. Gónzalez-Fernández, Agroindustrial waste as a resource for volatile fatty acids production via anaerobic fermentation, Bioresour. Technol. 297 (2020) 122486, https://doi.org/10.1016/j.biortech.2019.122486.
[41] N. Bermúdez-Penabad, C. Kennes, M.C. Veiga, Anaerobic digestion of tuna waste for the production of volatile fatty acids, Waste Manag 68 (2017) 96–102, https://doi.org/10.1016/j.wasman.2017.06.010.
[42] G. Strazzera, F. Battista, M. Andreolli, M. Menini, D. Bolzonella, S. Lampis, Influence of different household Food Wastes Fractions on Volatile Fatty Acids production by anaerobic fermentation, Bioresour. Technol. 335 (2021) 125289, https://doi.org/10.1016/j.biortech.2021.125289.
[43] K. Wang, J. Yin, D. Shen, N. Li, Anaerobic digestion of food waste for volatile fatty acids (VFAs) production with different types of inoculum: Effect of pH, Bioresour. Technol. 161 (2014) 395–401, https://doi.org/10.1016/j.biortech.2014.03.088.
[44] D. Zhang, X. Li, S. Jia, L. Dai, J. Zhao, Y. Chen, X. Dai, A review: factors affecting excess sludge anaerobic digestion for volatile fatty acids production, Water Sci. Technol. 72 (2015) 678–688, https://doi.org/10.2166/wst.2015.280.
[45] J. Garcia-Aguirre, E. Aymerich, J. González-Mtnez. de Goñi, M. Esteban-Gutiérrez, Selective VFA production potential from organic waste streams: Assessing temperature and pH influence, Bioresour. Technol. 244 (2017) 1081–1088, https://doi.org/10.1016/j.biortech.2017.07.187.
[46] Q. Fang, S. Ji, D. Huang, Z. Huang, Z. Huang, Y. Zeng, Y. Liu, Impact of alkaline pretreatment to enhance volatile fatty acids (VFAs) production from rice husk, Biochem. Res. Int. 2019 (2019) 8489747, https://doi.org/10.1155/2019/8489747.
[47] I. Owusu-Agyeman, E. Plaza, Z. Cetecioglu, Long-term alkaline volatile fatty acids production from waste streams: impact of pH and dominance of *dysgonomonadaceae*, Bioresour. Technol. 346 (2022) 126621, https://doi.org/10.1016/j.biortech.2021.126621.
[48] M. Ma, D. Zeng, X. Huang, Y. Mao, C. Liu, H. Wang, Continuous alkaline fermentation with peroxydisulfate pretreatment of dewatered sludge: microbial mechanisms for doubling the production of volatile fatty acids, ACS EST Water 5 (2025) 6853–6865, https://doi.org/10.1021/acsestwater.5c00797.
[49] A.S.S. Pinto, L.J. McDonald, R.J. Jones, J. Massanet-Nicolau, A. Guwy, M. McManus, Production of volatile fatty acids by anaerobic digestion of biowastes: Techno-economic and life cycle assessments, Bioresour. Technol. 388 (2023) 129726, https://doi.org/10.1016/j.biortech.2023.129726.
[50] A.F.A. Rahim, S.R.M. Kutty, E.H. Ezechi, Volatile Fatty Acids Production through Degradation of Biomass by Anaerobic Digestion (Mesophilic and Thermophilic), Appl. Mech. Mater. 567 (2014) 172–176, https://doi.org/10.4028/www.scientific.net/AMM.567.172.
[51] M. Mihajlovic, K. Shahbaz, C. Vaneeckhaute, S. Baroutian, Hydrothermal processing for plastic waste valorisation: Technical pathways, environmental performance, and prospects for commercialisation, J. Environ. Chem. Eng. 13 (2025) 118966, https://doi.org/10.1016/j.jece.2025.118966.
[52] D. Liakos, G. Altiparmaki, S. Malamis, S. Vakalis, Hydrothermal Liquefaction for Biofuel Synthesis: Assessment of VFA (Volatile Fatty Acid) and FAME (Fatty Acid Methyl Ester) Profiles from Spent Coffee Grounds, Energies 18 (2025) 2094, https://doi.org/10.3390/en18082094.
[53] A. Mathanker, S. Das, D. Pudasainee, M. Khan, A. Kumar, R. Gupta, A Review of Hydrothermal Liquefaction of Biomass for Biofuels Production with a Special

Focus on the Effect of Process Parameters, Co-Solvents, and Extraction Solvents, Energies 14 (2021) 4916, https://doi.org/10.3390/en14164916.
[54] T.H. Pedersen, L.A. Rosendahl, Production of fuel range oxygenates by supercritical hydrothermal liquefaction of lignocellulosic model systems, Biomass Bioenergy 83 (2015) 206–215, https://doi.org/10.1016/j.biombioe.2015.09.014.
[55] B.E. Eboibi, O. Eboibi, O.L. Okan, E.C. Udochukwu, P.E. Uku, S.E. Agarry, Hydrothermal liquefaction of microalga with and without seawater: effects of reaction temperature on yield and hydrocarbon species distribution in biocrude, Environ. Prog. Sustain. Energy 43 (2024) e14440, https://doi.org/10.1002/ep.14440.
[56] Y. Xu, M.F. Li, Hydrothermal liquefaction of lignocellulose for value-added products: Mechanism, parameter and production application, Bioresour. Technol. 342 (2021) 126035, https://doi.org/10.1016/j.biortech.2021.126035.
[57] A.S. Jatoi, A.A. Shah, J. Ahmed, S. Rehman, S.H. Sultan, A.K. Shah, A. Raza, N. M. Mubarak, Z. Hashmi, M.A. Usto, M. Murtaza, Hydrothermal liquefaction of lignocellulosic and protein-containing biomass: a comprehensive review, Catalysts 12 (2022) 1621, https://doi.org/10.3390/catal12121621.
[58] R.E. Ramos, M.C. Márquez, Volatile fatty acids from household food waste: production and kinetics, ChemEngineering 8 (2024) 84, https://doi.org/10.3390/chemengineering8050084.
[59] J.S. Passos, C. Lorentz, D. Laurenti, S.-J. Royer, I. Chontzoglou, P. Biller, Hydrothermal liquefaction of plastic marine debris from the North Pacific Garbage Patch, Resour. Conserv. Recycl 209 (2024) 107822, https://doi.org/10.1016/j.resconrec.2024.107822.
[60] K. Prapaiwatcharapan, S. Sunphorka, P. Kuchonthara, K. Kangvansaichol, N. Hinchiranan, Single- and two-step hydrothermal liquefaction of microalgae in a semi-continuous reactor: Effect of the operating parameters, Bioresour. Technol. 191 (2015) 426–432, https://doi.org/10.1016/j.biortech.2015.04.027.
[61] M.J. Boel, H. Wang, A.A. Farra, L. Megido, J.M. González-LaFuente, N.R. Shiju, Hydrothermal liquefaction of plastics: a survey of the effect of reaction conditions on the reaction efficiency, React. Chem. Eng. 9 (2024) 1014–1031, https://doi.org/10.1039/D2RE00510G.
[62] A. Yousefifar, S. Baroutian, M.M. Farid, D.J. Gapes, B.R. Young, Fundamental mechanisms and reactions in non-catalytic subcritical hydrothermal processes: A review, Water Res 123 (2017) 607–622, https://doi.org/10.1016/j.watres.2017.06.069.
[63] S.K. Bhargava, J. Tardio, J. Prasad, K. Föger, D.B. Akolekar, S.C. Grocott, Wet oxidation and catalytic wet oxidation, Ind. Eng. Chem. Res. 45 (2006) 1221–1258, https://doi.org/10.1021/ie051059n.
[64] Y. Khan, G.K. Anderson, D.J. Elliott, Wet oxidation of activated sludge, Water Res 33 (1999) 1681–1687, https://doi.org/10.1016/S0043-1354(98)00387-X.
[65] K. Fang, Y. Tong, G. Yao, N. Zhu, L. Jin, Y. Zhou, J. Zhao, NaOH-enhanced wet air oxidation of municipal sludge for high-quality carbon source production, Processes 13 (2025) 341, https://doi.org/10.3390/pr13020341.
[66] E. Krisner, M. Ambrosio, C. Massiani, Wet air oxidation of solid waste made of polymers, J. Environ. Eng. 126 (2000) 289–292, https://doi.org/10.1061/(ASCE)0733-9372(2000)126:3(289).
[67] H. Mumtaz, S. Werle, S. Sobek, A waste wet oxidation technique as a solution for chemical production and resource recovery in Poland, Clean. Technol. Environ. Policy 26 (2024) 1363–1382, https://doi.org/10.1007/s10098-023-02520-4.
[68] D. Anthraper, J. McLaren, S. Baroutian, M.T. Munir, B.R. Young, Hydrothermal deconstruction of municipal solid waste for solid reduction and value production, J. Clean. Prod. 201 (2018) 812–819, https://doi.org/10.1016/j.jclepro.2018.08.116.
[69] F. Javid, X. Xin, J.G.B. Derraik, W.A. Anderson, Y. Anderson, S. Baroutian, Hydrothermal deconstruction of single-use personal protective equipment during the COVID-19 pandemic, Waste Manag 153 (2022) 178–187, https://doi.org/10.1016/j.wasman.2022.09.006.
[70] C. González, M.I. Pariente, R. Molina, L.G. Espina, M.O. Masa, V. Bernal, J. A. Melero, F. Martínez, Increasing biodegradability of a real amine-contaminated spent caustic problematic stream through WAO and CWAO oxidation using a high specific surface catalyst from petcoke, Chem. Eng. J. 460 (2023) 141692, https://doi.org/10.1016/j.cej.2023.141692.
[71] Y.-K. Cheah, C. Vidal-Antich, J. Dosta, J. Mata-Álvarez, Volatile fatty acid production from mesophilic acidogenic fermentation of organic fraction of municipal solid waste and food waste under acidic and alkaline pH, Environ. Sci. Pollut. Res. 26 (2019) 35509–35522, https://doi.org/10.1007/s11356-019-05394-6.
[72] F. Yu, N. Li, H. Li, Y. Zhang, L. Chen, B. Wang, X. Sheng, J. Zhang, Q. Ping, H. Xiao, Boosting volatile fatty acids (VFAs) production during anaerobic digestion of rich lignocellulose pulp mill excess sludge by inoculating rumen fluid and the study on the microbial community structures and functions, Int. J. Biol. Macromol. 303 (2025) 140718, https://doi.org/10.1016/j.ijbiomac.2025.140718.
[73] Y.A. da Fonseca, A.B. de Camargos, G.S.M. Gomes, P. Fregulia, S.Q. Silva, L.V. A. Gurgel, B.E.L. Baêta, Enhancing biobased volatile fatty acids production from olive mill solid waste by optimization of pH and substrate to inoculum ratio, Processes 11 (2023) 338, https://doi.org/10.3390/pr11020338.
[74] E. Reyhanitash, B. Zaalberg, S.R.A. Kersten, B. Schuur, Extraction of volatile fatty acids from fermented wastewater, Sep. Purif. Technol. 161 (2016) 61–68, https://doi.org/10.1016/j.seppur.2016.01.037.
[75] H.M. Haflich, J.W. Singleton, O. Coronell, Relative contributions of mobility and partitioning to volatile fatty acid flux during electrodialysis, J. Membr. Sci. 711 (2024) 123204, https://doi.org/10.1016/j.memsci.2024.123204.
[76] T.H. Duong, T.T.V. Nga, Production of Volatile Fatty Acids from Organic-Rich Waste Streams: Current Issues, Challenges, and Opportunities, Curr. Pollut. Rep. 10 (2024) 594–605, https://doi.org/10.1007/s40726-024-00321-x.
[77] M. Atasoy, I. Owusu-Agyeman, E. Plaza, Z. Cetecioglu, Bio-based volatile fatty acid production and recovery from waste streams: Current status and future challenges, Bioresour. Technol. 268 (2018) 773–786, https://doi.org/10.1016/j.biortech.2018.07.042.
[78] Š. Schlosser, R. Kertész, J. Marták, Recovery and separation of organic acids by membrane-based solvent extraction and pertraction: an overview with a case study on recovery of MPCA, Sep. Purif. Technol. 41 (2005) 237–266, https://doi.org/10.1016/j.seppur.2004.07.019.
[79] C. Torri, C. Samorì, V. Ajao, S. Baraldi, P. Galletti, E. Tagliavini, Pertraction of volatile fatty acids through biodiesel-based liquid membranes, Chem. Eng. J. 366 (2019) 254–263, https://doi.org/10.1016/j.cej.2019.02.081.
[80] N.M. Farhan, S.S. Ibrahim, Q.F. Alsalhy, Modeling and simulation of pervaporation (PV) separation for alcohol dehydration, Heliyon 9 (2023), https://doi.org/10.1016/j.heliyon.2023.e13713.
[81] A. Chaudhuri, B.M. Harahap, B.K. Ahring, Membrane Technologies for Separating Volatile Fatty Acids Produced Through Arrested Anaerobic Digestion: A Review, Clean. Technol. 7 (2025) 48, https://doi.org/10.3390/cleantechnol7020048.
[82] P. Arana-Agudelo, M. Moussa, I.-C. Trelea, K. Lachin, V. Athès, Reactive liquid–liquid extraction of bio-based 3-hydroxypropionic acid using a biocompatible organic phase containing a tertiary amine: A model-based approach to elucidate dominant mechanisms, Sep. Purif. Technol. 294 (2022) 121184, https://doi.org/10.1016/j.seppur.2022.121184.
[83] L.M.J. Sprakel, B. Schuur, Solvent developments for liquid-liquid extraction of carboxylic acids in perspective, Sep. Purif. Technol. 211 (2019) 935–957, https://doi.org/10.1016/j.seppur.2018.10.023.
[84] V. Outram, Y. Zhang, Solvent-free membrane extraction of volatile fatty acids from acidogenic fermentation, Bioresour. Technol. 270 (2018) 400–408, https://doi.org/10.1016/j.biortech.2018.09.057.
[85] J.T. Dafoe, A.J. Daugulis, situ product removal in fermentation systems: improved process performance and rational extractant selection, Biotechnol. Lett. 36 (2014) 443–460, https://doi.org/10.1007/s10529-013-1380-6.
[86] L. Shi, Y. Hu, S. Xie, G. Wu, Z. Hu, X. Zhan, Recovery of nutrients and volatile fatty acids from pig manure hydrolysate using two-stage bipolar membrane electrodialysis, Chem. Eng. J. 334 (2018) 134–142, https://doi.org/10.1016/j.cej.2017.10.010.
[87] J.S. Jaime-Ferrer, E. Couallier, Ph Viers, G. Durand, M. Rakib, Three-compartment bipolar membrane electrodialysis for splitting of sodium formate into formic acid and sodium hydroxide: role of diffusion of molecular acid, J. Membr. Sci. 325 (2008) 528–536, https://doi.org/10.1016/j.memsci.2008.07.059.
[88] S. Rebecchi, D. Pinelli, L. Bertin, F. Zama, F. Fava, D. Frascari, Volatile fatty acids recovery from the effluent of an acidogenic digestion process fed with grape pomace by adsorption on ion exchange resins, Chem. Eng. J. 306 (2016) 629–639, https://doi.org/10.1016/j.cej.2016.07.101.
[89] E. Reyhanitash, S.R.A. Kersten, B. Schuur, Recovery of volatile fatty acids from fermented wastewater by adsorption, ACS Sustain. Chem. Eng. 5 (2017) 9176–9184, https://doi.org/10.1021/acssuschemeng.7b02095.
[90] T. Xing, S. Yu, J. Tang, H. Liu, F. Zhen, Y. Sun, X. Kong, Liquid–liquid extraction of volatile fatty acids from anaerobic acidification broth using ionic liquids and cosolvent, Energies 16 (2023) 785, https://doi.org/10.3390/en16020785.
[91] J.M. Dias, A. Oehmen, L.S. Serafim, P.C. Lemos, M.A. Reis, R. Oliveira, Metabolic modelling of polyhydroxyalkanoate copolymers production by mixed microbial cultures, BMC Syst. Biol. 2 (2008) 59, https://doi.org/10.1186/1752-0509-2-59.
[92] R. Paduvari, D.M. Somashekara, Advancements in genetic engineering for enhanced Polyhydroxyalkanoates (PHA) production: a comprehensive review of metabolic pathway manipulation and gene deletion strategies, Bioengineered 16 (2025) 2458363, https://doi.org/10.1080/21655979.2025.2458363.
[93] D. Vicente, D.N. Proença, P.V. Morais, The Role of Bacterial Polyhydroalkanoate (PHA) in a Sustainable Future: A Review on the Biological Diversity, Int. J. Environ. Res. Public. Health 20 (2023) 2959, https://doi.org/10.3390/ijerph20042959.
[94] W. Blunt, R. Sparling, D. Gapes, D. Levin, N. Cicek, The role of dissolved oxygen content as a modulator of microbial polyhydroxyalkanoate synthesis, World J. Microbiol. Biotechnol. 34 (2018), https://doi.org/10.1007/s11274-018-2488-6.
[95] H.S. Kusuma, A. Sabita, N.A. Putri, N. Azliza, N. Illiyanasafa, H. Darmokoesoemo, A.N. Amenaghawon, T.A. Kurniawan, Waste to wealth: Polyhydroxyalkanoates (PHA) production from food waste for a sustainable packaging paradigm, Food Chem. Mol. Sci. 9 (2024) 100225, https://doi.org/10.1016/j.fochms.2024.100225.
[96] J.H. Yun, S.S. Sawant, B.S. Kim, Production of polyhydroxyalkanoates by Ralstonia eutropha from volatile fatty acids, Korean J. Chem. Eng. 30 (2013) 2223–2227, https://doi.org/10.1007/s11814-013-0190-9.
[97] N. Hernández-Herreros, A. Rodriguez, V. Rivero-Buceta, A. Rojas, M.A. Prieto, Flexible feeding strategy for high-yield PHA bioprocessing in *cupriavidus necator* H16 from anaerobically fermented industrial wastewater, Bioresour. Technol. 434 (2025) 132774, https://doi.org/10.1016/j.biortech.2025.132774.
[98] A. Ferre-Guell, J. Winterburn, Increased production of polyhydroxyalkanoates with controllable composition and consistent material properties by fed-batch fermentation, Biochem. Eng. J. 141 (2019) 35–42, https://doi.org/10.1016/j.bej.2018.10.004.
[99] G.W. Haywood, A.J. Anderson, D. Roger Williams, E.A. Dawes, D.F. Ewing, Accumulation of a poly(hydroxyalkanoate) copolymer containing primarily 3-hydroxyvalerate from simple carbohydrate substrates by *Rhodococcus* sp. NCIMB

40126, Int. J. Biol. Macromol. 13 (1991) 83–88, https://doi.org/10.1016/0141-8130(91)90053-W.
[100] I. Belyamani, K. Abdelghafar, M. Plouzeau, L. Najemi, K. Askar, L. Cauret, Comparative Analysis of Mechanical Recycling Simulation of Poly (Hydroxybutyrate-Co-Hydroxyvalerate) (PHBV): Injection Molding Vs. Extrusion Processes, J. Polym. Sci. n/a (n.d.). https://doi.org/10.1002/pol.20250603.
[101] K. Tajima, T. Igari, D. Nishimura, M. Nakamura, Y. Satoh, M. Munekata, Isolation and characterization of*Bacillus* sp. INT005 accumulating polyhydroxyalkanoate (PHA) from gas field soil, J. Biosci. Bioeng. 95 (2003) 77–81, https://doi.org/10.1016/S1389-1723(03)80152-4.
[102] S. Munir, N. Jamil, Polyhydroxyalkanoates (PHA) production in bacterial co-culture using glucose and volatile fatty acids as carbon source, J. Basic Microbiol 58 (2018) 247–254, https://doi.org/10.1002/jobm.201700276.
[103] F. Cerrone, S.K. Choudhari, R. Davis, D. Cysneiros, V. O'Flaherty, G. Duane, E. Casey, M.W. Guzik, S.T. Kenny, R.P. Babu, K. O'Connor, Medium chain length polyhydroxyalkanoate (mcl-PHA) production from volatile fatty acids derived from the anaerobic digestion of grass, Appl. Microbiol. Biotechnol. 98 (2014) 611–620, https://doi.org/10.1007/s00253-013-5323-x.
[104] K. Johnson, Y. Jiang, R. Kleerebezem, G. Muyzer, M.C.M. van Loosdrecht, Enrichment of a Mixed Bacterial Culture with a High Polyhydroxyalkanoate Storage Capacity, Biomacromolecules 10 (2009) 670–676, https://doi.org/10.1021/bm8013796.
[105] M.G.E. Albuquerque, V. Martino, E. Pollet, L. Avérous, M.A.M. Reis, Mixed culture polyhydroxyalkanoate (PHA) production from volatile fatty acid (VFA)-rich streams: Effect of substrate composition and feeding regime on PHA productivity, composition and properties, J. Biotechnol. 151 (2011) 66–76, https://doi.org/10.1016/j.jbiotec.2010.10.070.
[106] G. Du, Y. Si, J. Yu, Inhibitory effect of medium-chain-length fatty acids on synthesis of polyhydroxyalkanoates from volatile fatty acids by Ralstonia eutropha, Biotechnol. Lett. 23 (2001) 1613–1617, https://doi.org/10.1023/A:1011916131544.
[107] K. Wang, R. Zhang, Production of polyhydroxyalkanoates (PHA) by haloferax mediterranei from food waste derived nutrients for biodegradable plastic applications, J. Microbiol. Biotechnol. 31 (2021) 338–347, https://doi.org/10.4014/jmb.2008.08057.
[108] M. Parroquin-Gonzalez, J. Winterburn, Continuous bioreactor production of polyhydroxyalkanoates in haloferax mediterranei, Front. Bioeng. Biotechnol. 11 (2023), https://doi.org/10.3389/fbioe.2023.1220271.
[109] T. Dan, H. Jing, T. Shen, J. Zhu, Y. Liu, Performance of production of polyhydroxyalkanoates from food waste fermentation with *rhodopseudomonas palustris*, Bioresour. Technol. 385 (2023) 129165, https://doi.org/10.1016/j.biortech.2023.129165.
[110] F. Cai, M. Lin, W. Jin, C. Chen, G. Liu, Biosynthesis of poly(3-hydroxybutyrate-co-3-hydroxvalerate) from volatile fatty acids by cupriavidus necator, J. Basic Microbiol 63 (2023) 128–139, https://doi.org/10.1002/jobm.202200448.
[111] D.H. Vu, A. Mahboubi, A. Root, I. Heinmaa, M.J. Taherzadeh, D. Åkesson, Thorough Investigation of the Effects of Cultivation Factors on Polyhydroalkanoates (PHAs) Production by Cupriavidus necator from Food Waste-Derived Volatile Fatty Acids, Fermentation 8 (2022) 605, https://doi.org/10.3390/fermentation8110605.
[112] Q. Wen, Z. Chen, T. Tian, W. Chen, Effects of phosphorus and nitrogen limitation on PHA production in activated sludge, J. Environ. Sci. 22 (2010) 1602–1607, https://doi.org/10.1016/S1001-0742(09)60295-3.
[113] S. Wijeyekoon, C.R. Carere, M. West, S. Nath, D. Gapes, Mixed culture polyhydroxyalkanoate (PHA) synthesis from nutrient rich wet oxidation liquors, Water Res 140 (2018) 1–11, https://doi.org/10.1016/j.watres.2018.04.017.
[114] K. Mohanrasu, R.G.R. Rao, G.H. Dinesh, K. Zhang, G.S. Prakash, D.-P. Song, S. Muniyasamy, A. Pugazhendhi, J. Jeyakanthan, A. Arun, Optimization of media components and culture conditions for polyhydroxyalkanoates production by *Bacillus megaterium*, Fuel 271 (2020) 117522, https://doi.org/10.1016/j.fuel.2020.117522.
[115] O.A. Odeniyi, O.J. Adeola, Production and characterization of polyhydroxyalkanoic acid from *Bacillus thuringiensis* using different carbon substrates, Int. J. Biol. Macromol. 104 (2017) 407–413, https://doi.org/10.1016/j.ijbiomac.2017.06.041.
[116] P. Luangthongkam, P.J. Strong, S.N. Syed Mahamud, P. Evans, P. Jensen, G. Tyson, B. Laycock, P.A. Lant, S. Pratt, The effect of methane and odd-chain fatty acids on 3-hydroxybutyrate (3HB) and 3-hydroxyvalerate (3HV) synthesis by a methylosinus-dominated mixed culture, Bioresour. Bioprocess 6 (2019) 50, https://doi.org/10.1186/s40643-019-0285-1.
[117] A. Jin, L.J. del Valle, J. Puiggalí, Copolymers and blends based on 3-hydroxybutyrate and 3-hydroxyvalerate units, Int. J. Mol. Sci. 24 (2023) 17250, https://doi.org/10.3390/ijms242417250.
[118] K. Chotchindakun, W. Pathom-Aree, K. Dumri, J. Ruangsuriya, C. Pumas, J. Pekkoh, Low crystallinity of poly(3-hydroxybutyrate-co-3-hydroxyvalerate) bioproduction by hot spring cyanobacterium cyanosarcina sp. AARL T020, Plants 10 (2021) 503, https://doi.org/10.3390/plants10030503.
[119] A.L.M. Martínez, M. Yepes-Pérez, K.A.C. Contreras, P.E.Z. Moreno, Production of poly(3-hydroxybutyrate-Co-3-hydroxyvalerate) by bacillus megaterium LVN01 using biogas digestate, Appl. Microbiol 4 (2024) 1057–1078, https://doi.org/10.3390/applmicrobiol4030072.
[120] C. Amabile, T. Abate, S. Chianese, D. Musmarra, R. Muñoz, The co-conversion of methane and mixtures of volatile fatty acids into poly(3-hydroxybutyrate-co-3-hydroxyvalerate) expands the potential of an integrated biorefinery, Bioresour. Technol. 387 (2023) 129699, https://doi.org/10.1016/j.biortech.2023.129699.
[121] S. Puig, M. Coma, H. Monclús, M.C.M. van Loosdrecht, J. Colprim, M.D. Balaguer, Selection between alcohols and volatile fatty acids as external carbon sources for EBPR, Water Res 42 (2008) 557–566, https://doi.org/10.1016/j.watres.2007.07.050.
[122] D.H. Vu, S. Wainaina, M.J. Taherzadeh, D. Åkesson, J.A. Ferreira, Production of polyhydroxyalkanoates (PHAs) by Bacillus megaterium using food waste acidogenic fermentation-derived volatile fatty acids, Bioengineered 12 (2021) 2480–2498, https://doi.org/10.1080/21655979.2021.1935524.
[123] Y. Park, J.-M. Jeon, J.-K. Park, Y.-H. Yang, S.S. Choi, J.-J. Yoon, Optimization of polyhydroxyalkanoate production in halomonas sp. YLGW01 using mixed volatile fatty acids: a study on mixture analysis and fed-batch strategy, Microb. Cell Factor. 22 (2023) 171, https://doi.org/10.1186/s12934-023-02188-x.
[124] M.S. Duarte, J.V. Oliveira, C. Pereira, M. Carvalho, D.P. Mesquita, M.M. Alves, Volatile fatty acids (VFA) production from wastewaters with high salinity—influence of pH, salinity and reactor configuration, Fermentation 7 (2021) 303, https://doi.org/10.3390/fermentation7040303.
[125] F. Huschner, E. Grousseau, C.J. Brigham, J. Plassmeier, M. Popovic, C. Rha, A. J. Sinskey, Development of a feeding strategy for high cell and PHA density fed-batch fermentation of *ralstonia eutropha* H16 from organic acids and their salts, Process Biochem 50 (2015) 165–172, https://doi.org/10.1016/j.procbio.2014.12.004.
[126] L. Garcia-Gonzalez, H. De Wever, Acetic acid as an indirect sink of CO2 for the synthesis of polyhydroxyalkanoates (PHA): comparison with PHA production processes directly using CO2 as feedstock, Appl. Sci. 8 (2018) 1416, https://doi.org/10.3390/app8091416.
[127] R. Lo Coco, A. Jelic, E. Järvelä, N. Frison, Recovery of bio-based volatile fatty acids from anaerobically treated winery wastewater using a closed-loop liquid-liquid hydrophobic membrane contactor system, Chem. Eng. J. 500 (2024) 156889, https://doi.org/10.1016/j.cej.2024.156889.
[128] K.S. Barros, B.C. Marreiros, M.A.M. Reis, J.G. Crespo, V. Pérez-Herranz, S. Velizarov, Recovery and fractionation of volatile fatty acids from fermented solutions by electrodialysis: electrochemical characterization of anion-exchange membranes, J. Environ. Chem. Eng. 12 (2024) 114457, https://doi.org/10.1016/j.jece.2024.114457.
[129] Z. Chen, L. Zhao, Y. Ji, Q. Wen, L. Huang, Reconsideration on the effect of nitrogen on mixed culture polyhydroxyalkanoate production toward high organic loading enrichment history, Front. Environ. Sci. Eng. 13 (2019) 54, https://doi.org/10.1007/s11783-019-1135-2.
[130] I. Ntaikou, I. Koumelis, M. Kamilari, Z. Iatridi, C. Tsitsilianis, G. Lyberatos, Effect of nitrogen limitation on polyhydroxyalkanoates production efficiency, properties and microbial dynamics using a soil-derived mixed continuous culture, Int. J. Biobased Plast. 1 (2019) 31–47, https://doi.org/10.1080/24759651.2019.1648016.
[131] C. Kourmentza, J. Plácido, N. Venetsaneas, A. Burniol-Figols, C. Varrone, H. N. Gavala, M.A.M. Reis, Recent advances and challenges towards sustainable polyhydroxyalkanoate (PHA) production, Bioengineering 4 (2017) 55, https://doi.org/10.3390/bioengineering4020055.
[132] M. Bolla, M. Pettinato, P.F. Ferrari, B. Fabiano, P. Perego, Polyhydroxyalkanoates production from laboratory to industrial scale: A review, Int. J. Biol. Macromol. 310 (2025) 143255, https://doi.org/10.1016/j.ijbiomac.2025.143255.
[133] S. Mohammed, H.T. Behera, A. Dekebo, L. Ray, Optimization of the culture conditions for production of Polyhydroxyalkanoate and its characterization from a new *Bacillus cereus* sp. *BNPI*-92 strain, isolated from plastic waste dumping yard, Int. J. Biol. Macromol. 156 (2020) 1064–1080, https://doi.org/10.1016/j.ijbiomac.2019.11.138.
[134] M. Villano, M. Beccari, D. Dionisi, S. Lampis, A. Miccheli, G. Vallini, M. Majone, Effect of pH on the production of bacterial polyhydroxyalkanoates by mixed cultures enriched under periodic feeding, Process Biochem 45 (2010) 714–723, https://doi.org/10.1016/j.procbio.2010.01.008.
[135] M.S. Munna, Z. Zeba, R. Noor, Influence of temperature on the growth of pseudomonas putida, Stamford J. Microbiol 5 (2015) 9–12, https://doi.org/10.3329/sjm.v5i1.26912.
[136] P.C. Sabapathy, S. Devaraj, A. Parthipan, P. Kathirvel, Polyhydroxyalkanoate production from statistically optimized media using rice mill effluent as sustainable substrate with an analysis on the biopolymer's degradation potential, Int. J. Biol. Macromol. 126 (2019) 977–986, https://doi.org/10.1016/j.ijbiomac.2019.01.003.
[137] R. Rathika, V. Janaki, K. Shanthi, S. Kamala-Kannan, Bioconversion of agro-industrial effluents for polyhydroxyalkanoates production using Bacillus subtilis RS1, Int. J. Environ. Sci. Technol. 16 (2019) 5725–5734, https://doi.org/10.1007/s13762-018-2155-3.
[138] A.V. Samrot, S.K. Samanvitha, N. Shobana, E.R. Renitta, P. Senthilkumar, S. S. Kumar, S. Abirami, S. Dhiva, M. Bavanilatha, P. Prakash, S. Saigeetha, K. S. Shree, R. Thirumurugan, The synthesis, characterization and applications of polyhydroxyalkanoates (PHAs) and PHA-based nanoparticles, Polymers 13 (2021), https://doi.org/10.3390/polym13193302.
[139] T. Mandragutti, T.S. Jarso, S. Godi, S.S. Begum, B. K, Physicochemical characterization of polyhydroxybutyrate (PHB) produced by the rare halophile brachybacterium paraconglomeratum MTCC 13074, Microb. Cell Factor. 23 (2024) 59, https://doi.org/10.1186/s12934-024-02324-1.
[140] S. Maity, S. Das, S. Mohapatra, A.D. Tripathi, J. Akthar, S. Pati, S. Pattnaik, D. P. Samantaray, Growth associated polyhydroxybutyrate production by the novel *Zobellellae tiwanensis* strain DD5 from banana peels under submerged fermentation, Int. J. Biol. Macromol. 153 (2020) 461–469, https://doi.org/10.1016/j.ijbiomac.2020.03.004.

[141] T. Rakkan, N. Chana, K. Sangkharak, The integration of textile wastewater treatment with polyhydroxyalkanoate production using newly isolated enterobacter strain TS3, Waste Biomass-.-. Valoriz. 13 (2022) 571–582, https://doi.org/10.1007/s12649-021-01504-z.
[142] T. Hahn, M.O. Alzate, S. Leonhardt, P. Tamang, S. Zibek, Current trends in medium-chain-length polyhydroxyalkanoates: microbial production, purification, and characterization, Eng. Life Sci. 24 (2024) 2300211, https://doi.org/10.1002/elsc.202300211.
[143] S. Mohapatra, B. Sarkar, D.P. Samantaray, A. Daware, S. Maity, S. Pattnaik, S. Bhattacharjee, Bioconversion of fish solid waste into PHB using bacillus subtilis based submerged fermentation process, Environ. Technol. 38 (2017) 3201–3208, https://doi.org/10.1080/09593330.2017.1291759.
[144] K. Ishida, N. Asakawa, Y. Inoue, Structure, properties and biodegradation of some bacterial copoly(hydroxyalkanoate)s, Macromol. Symp. 224 (2005) 47–58, https://doi.org/10.1002/masy.200550605.
[145] B. Laycock, P. Halley, S. Pratt, A. Werker, P. Lant, The chemomechanical properties of microbial polyhydroxyalkanoates, Prog. Polym. Sci. 39 (2014) 397–442, https://doi.org/10.1016/j.progpolymsci.2013.06.008.
[146] L. Feng, J. Yan, Z. Jiang, X. Chen, Z. Li, J. Liu, X. Qian, Z. Liu, G. Liu, C. Liu, Y. Wang, G. Hu, W. Dong, Z. Cui, Characterization of polyhydroxybutyrate (PHB) synthesized by newly isolated rare actinomycetes *Aquabacterium* sp. A7-Y, Int. J. Biol. Macromol. 232 (2023) 123366, https://doi.org/10.1016/j.ijbiomac.2023.123366.
[147] F. Ridella, E. Fernández, S. Álvarez-García, I. Marcet, M. Rendueles, M. Díaz, Polyhydroxyalkanoates production from cocoa bean shell fat: bioprocess optimization and polymer characterization, Int. J. Biol. Macromol. 331 (2025) 148407, https://doi.org/10.1016/j.ijbiomac.2025.148407.
[148] A. Werker, R. Pei, K. Kim, G. Moretto, A. Estevez-Alonso, C. Vermeer, M. Arcos-Hernandez, J. Dijkstra, E. de Vries, Thermal pre-processing before extraction of polyhydroxyalkanoates for molecular weight quality control, Polym. Degrad. Stab. 209 (2023) 110277, https://doi.org/10.1016/j.polymdegradstab.2023.110277.
[149] R.D. Ashby, T.A. Foglia, Poly(hydroxyalkanoate) biosynthesis from triglyceride substrates, Appl. Microbiol. Biotechnol. 49 (1998) 431–437, https://doi.org/10.1007/s002530051194.
[150] T.A.T. Yasim-Anuar, M.N.F. Norrrahim, S. m Sapuan, R. a Ilyas, M.A. Jenol, N.A. M. Razali, M.I. Hakimi, N.F.A. Rahim, S.U.F.S. Najmuddin, Polyhydroxyalkanoates for packaging application. Bio-Based Packag, John Wiley & Sons, Ltd, 2021, pp. 279–293, https://doi.org/10.1002/9781119381228.ch16.
[151] S. Rodriguez-Perez, A. Serrano, A.A. Pantión, B. Alonso-Fariñas, Challenges of scaling-up PHA production from waste streams. A review, J. Environ. Manag. 205 (2018) 215–230, https://doi.org/10.1016/j.jenvman.2017.09.083.
[152] S. Kopf, D. Åkesson, M. Skrifvars, Textile fiber production of biopolymers – a review of spinning techniques for polyhydroxyalkanoates in biomedical applications, Polym. Rev. 63 (2023) 200–245, https://doi.org/10.1080/15583724.2022.2076693.
[153] Danimer Scientific, Versatile and sustainable, PHA can be used to replace a wide variety of traditional plastics, Versatile Sustain. PHA Can Be Used Replace Wide Var, Tradit. Plast. (2025). ⟨https://danimerscientific.com/applications-for-pha/⟩ (accessed October 23, 2025).
[154] J. Poole, Danimer Scientific upscales PHA biopolymer production in challenge to petrochemical plastics, ⟨Https://Www.Foodingredientsfirst.Com⟩ (2021). ⟨https://www.foodingredientsfirst.com/news/danimer-scientific-upscales-pha-biopolymer-production-in-challenge-to-petrochemical-plastics.html⟩ (accessed October 23, 2025).
[155] CJ Biomaterials, Revolutionizing Sustainable Biopolymer Production, Revolutionizing Sustain. Biopolym. Prod. (2025). ⟨https://cjbiomaterials.com/technology/pha-process/⟩ (accessed October 23, 2025).
[156] RWDC Industries, PHA is the Future of Sustainable Packaging, (2024). ⟨https://www.rwdc-industries.com/pha-is-the-future-of-sustainable-packaging⟩ (accessed October 23, 2025).
[157] Kaneka Corporation, Biodegradable polymer | Material Solutions Unit | Business | KANEKA CORPORATION, (2025). ⟨https://www.kaneka.co.jp/en/business/material/nbd_001.html⟩ (accessed October 23, 2025).
[158] C. Utsunomia, Q. Ren, M. Zinn, Poly(4-Hydroxybutyrate): Current State and Perspectives, Front. Bioeng. Biotechnol. 8 (2020), https://doi.org/10.3389/fbioe.2020.00257.
[159] Tepha Inc, Tepha Inc. Announces Regenerative Medicine Recent Developments and R & D Initiatives, (2017). ⟨https://www.prnewswire.com/news-releases/tepha-inc-announces-regenerative-medicine-recent-developments-and-r–d-initiatives-300551472.html⟩ (accessed October 23, 2025).
[160] M. Koller, A. Mukherjee, A New Wave of Industrialization of PHA Biopolyesters, Bioengineering 9 (2022) 74, https://doi.org/10.3390/bioengineering9020074.
[161] AIMPLAS, COM4PHA: new biodegradable bioplastics for cosmetics, food and agricultural packaging, (2025). ⟨https://www.aimplas.net/blog/com4pha-new-biodegradable-bioplastics-cosmetics-food-agricultural-packaging/⟩ (accessed October 23, 2025).
[162] D.C. Monagas, COM4PHA project explores bioplastic alternatives for packaging and agriculture, World Bio Mark. Insights (2025). ⟨https://worldbiomarketinsights.com/com4pha-project-explores-bioplastic-alternatives-for-packaging-and-agriculture/⟩ (accessed October 23, 2025).
[163] X. Tao, S. Liu, L. Lv, L. Sun, G. Zhang, J. Liang, W. Zou, Recent advances in polyhydroxyalkanoate production from volatile fatty acids derived from food waste fermentation, Front. Microbiol 16 (2025), https://doi.org/10.3389/fmicb.2025.1693596.
[164] Z. Zhang, Y. Lin, S. Wu, X. Li, J.J. Cheng, C. Yang, Effect of composition of volatile fatty acids on yield of polyhydroxyalkanoates and mechanisms of bioconversion from activated sludge, Bioresour. Technol. 385 (2023) 129445, https://doi.org/10.1016/j.biortech.2023.129445.
[165] R. Chandra, A. Thakor, T.H. Mekonnen, T.C. Charles, H.-S. Lee, Production of polyhydroxyalkanoate (PHA) copolymer from food waste using mixed culture for carboxylate production and *Pseudomonas putida* for PHA synthesis, J. Environ. Manag. 336 (2023) 117650, https://doi.org/10.1016/j.jenvman.2023.117650.
[166] K. Srirangan, X. Liu, T.T. Tran, T.C. Charles, M. Moo-Young, C.P. Chou, Engineering of Escherichia coli for direct and modulated biosynthesis of poly(3-hydroxybutyrate-co-3-hydroxyvalerate) copolymer using unrelated carbon sources, Sci. Rep. 6 (2016) 36470, https://doi.org/10.1038/srep36470.
[167] N.O. Zhila, K.Y. Sapozhnikova, E.G. Kiselev, A.D. Vasiliev, I.V. Nemtsev, E. I. Shishatskaya, T.G. Volova, Properties of Degradable Polyhydroxyalkanoates (PHAs) Synthesized by a New Strain, Cupriavidus necator IBP/SFU-1, from Various Carbon Sources, Polymers 13 (2021) 3142, https://doi.org/10.3390/polym13183142.
[168] E. Attenborough, F. Yazdan Parast, R. Nosrati, M.M. Banaszak Holl, L. van 't Hag, Bacterial species-structure-property relationships of polyhydroxyalkanoate biopolymers produced on simple sugars for thin film applications, Microb. Cell Factor. 24 (2025) 204, https://doi.org/10.1186/s12934-025-02833-7.
[169] A.K. Chandel, S.S. da Silva, O.V. Singh, Detoxification of lignocellulose hydrolysates: biochemical and metabolic engineering toward white biotechnology, Bioenergy Res 6 (2013) 388–401, https://doi.org/10.1007/s12155-012-9241-z.
[170] T.L. Richardson, N.K. Harner, P.K. Bajwa, J.T. Trevors, H. Lee, Approaches to deal with toxic inhibitors during fermentation of lignocellulosic substrates. Sustain. Prod. Fuels Chem. Fibers For. Biomass, American Chemical Society, 2011, pp. 171–202, https://doi.org/10.1021/bk-2011-1067.ch007.
[171] W. Parawira, M. Tekere, Biotechnological strategies to overcome inhibitors in lignocellulose hydrolysates for ethanol production: review, Crit. Rev. Biotechnol. 31 (2011) 20–31, https://doi.org/10.3109/07388551003757816.
[172] J. Chen, X. Zhang, R.D. Tyagi, P. Drogui, Utilization of methanol in crude glycerol to assist lipid production in non-sterilized fermentation from *trichosporon oleaginosus*, Bioresour. Technol. 253 (2018) 8–15, https://doi.org/10.1016/j.biortech.2018.01.008.
[173] A. Surendran, M. Lakshmanan, J.Y. Chee, A.M. Sulaiman, D.V. Thuoc, K. Sudesh, Can polyhydroxyalkanoates be produced efficiently from waste plant and animal oils? Front. Bioeng. Biotechnol. 8 (2020) 169, https://doi.org/10.3389/fbioe.2020.00169.
[174] G. Agustín Martinez, L. Bertin, A. Scoma, S. Rebecchi, G. Braunegg, F. Fava, Production of polyhydroxyalkanoates from dephenolised and fermented olive mill wastewaters by employing a pure culture of *Cupriavidus necator*, Biochem. Eng. J. 97 (2015) 92–100, https://doi.org/10.1016/j.bej.2015.02.015.
[175] J. Chen, W. Li, Z.-Z. Zhang, T.-W. Tan, Z.-J. Li, Metabolic engineering of Escherichia coli for the synthesis of polyhydroxyalkanoates using acetate as a main carbon source, Microb. Cell Factor. 17 (2018) 102, https://doi.org/10.1186/s12934-018-0949-0.
[176] A. Paloyan, M. Tadevosyan, D. Ghevondyan, L. Khoyetsyan, M. Karapetyan, A. Margaryan, G. Antranikian, H. Panosyan, Biodegradation of polyhydroxyalkanoates: current state and future prospects, Front. Microbiol 16 (2025) 1542468, https://doi.org/10.3389/fmicb.2025.1542468.
[177] M. Koller, D. Heeney, A. Mukherjee, Biodegradability of polyhydroxyalkanoate (PHA) biopolyesters in nature: a review, Biodegradation 36 (2025) 76, https://doi.org/10.1007/s10532-025-10164-y.
[178] S. Chegini, A.R.M. Sikkander, M. Masoudi, H. Ekhtari, E. Mojaver, H. Jafari, A circular bioeconomy framework for biodegradable waste: strategies and opportunities, Bioresour. Bioprod. 2 (2026) 2, https://doi.org/10.3390/bioresourbioprod2010002.
[179] E.B. de Figueiredo, A.R. Panosso, R. Romão, N. La Scala, Greenhouse gas emission associated with sugar production in southern Brazil, Carbon Balance Manag 5 (2010) 3, https://doi.org/10.1186/1750-0680-5-3.
[180] J.-L. Gálvez-Martos, S. Greses, J.-A. Magdalena, D. Iribarren, E. Tomás-Pejó, C. González-Fernández, Life cycle assessment of volatile fatty acids production from protein- and carbohydrate-rich organic wastes, Bioresour. Technol. 321 (2021) 124528, https://doi.org/10.1016/j.biortech.2020.124528.
[181] Y.K. Leong, P.L. Show, H.C. Lin, C.K. Chang, H.-S. Loh, J.C.-W. Lan, T.C. Ling, Preliminary integrated economic and environmental analysis of polyhydroxyalkanoates (PHAs) biosynthesis, Bioresour. Bioprocess 3 (2016) 41, https://doi.org/10.1186/s40643-016-0120-x.
[182] WBCSD, Circular Economy Action Plan (CEAP) 2020 summary for business, 2020. ⟨https://docs.wbcsd.org/2020/11/WBCSD_Circular_Economy_Action_Plan_2020%E2%80%93Summary_for_business.pdf⟩.
[183] M. Arriaga, F.J. Pinar, I. Izarra, J. del Amo, J. Vicente, F.J. Fernández-Morales, J. Mena, Valorization of agri-food waste into PHA and bioplastics: from waste selection to transformation, Appl. Sci. 15 (2025) 1008, https://doi.org/10.3390/app15031008.
[184] UN Environement Programme, Goal-12 | sustainable developments goals, (n.d.). ⟨https://sdgs.unep.org/article/goal-12⟩ (accessed January 29, 2026).
[185] M. Gundlapalli, S. Ganesan, Polyhydroxyalkanoates (PHAs): Key Challenges in production and sustainable strategies for cost reduction within a circular economy framework, Results Eng. 26 (2025) 105345, https://doi.org/10.1016/j.rineng.2025.105345.
[186] S. Vigneswari, M.S.M. Noor, T.S.M. Amelia, K. Balakrishnan, A. Adnan, K. Bhubalan, A.-A.A. Amirul, S. Ramakrishna, Recent Advances in the

Biosynthesis of Polyhydroxyalkanoates from Lignocellulosic Feedstocks, Life 11 (2021) 807, https://doi.org/10.3390/life11080807.
[187] M. Li, M.R. Wilkins, Recent advances in polyhydroxyalkanoate production: Feedstocks, strains and process developments, Int. J. Biol. Macromol. 156 (2020) 691–703, https://doi.org/10.1016/j.ijbiomac.2020.04.082.
[188] A.B. Ozturk, X. Kourilova, I. Buchtikova, S. Obruca, Techno-economic assessment of polyhydroxyalkanoates production from lignocellulosic biomass employing halophilic and thermophilic microbial platform: effect of fermentation conditions and downstream operations, Waste Manag 203 (2025) 114887, https://doi.org/10.1016/j.wasman.2025.114887.
[189] Ukhi India Pvt. Ltd, PHA market report 2025–2030, Polyhydroxyalkanoates PHA Mark. Size (2026). ⟨https://ukhi.com/ukhi-pha-market-report-2025-2030/⟩ (accessed April 11, 2026).
[190] M.I. Sepúlveda, M. Seeger, G. Vidal, Carbon recovery from wastewater feedstocks: synthesis of polyhydroxyalkanoates for target applications, Resources 14 (2025), https://doi.org/10.3390/resources14100156.
[191] R.F. Feito, A.J. Guwy, R.M. Dinsdale, J. Massanet-Nicolau, Real-time monitoring of volatile fatty acids using a novel VFAs analyser and automated high solids separation system, J. Water Process Eng. 76 (2025) 108157, https://doi.org/10.1016/j.jwpe.2025.108157.
[192] F. Pardelha, M.G.E. Albuquerque, M.A.M. Reis, R. Oliveira, J.M.L. Dias, Dynamic metabolic modelling of volatile fatty acids conversion to polyhydroxyalkanoates by a mixed microbial culture, N. Biotechnol. 31 (2014) 335–344, https://doi.org/10.1016/j.nbt.2013.06.008.
[193] X. Li, S. Wu, Y. Dong, H. Fan, Z. Bai, X. Zhuang, Engineering microbial consortia towards bioremediation, Water 13 (2021), https://doi.org/10.3390/w13202928.
[194] L. Pozo-Morales, A.R. Martínez, E. Baquerizo, G. del V. Agulla, Simulation tool for the techno-economic assessment of the integrated production of polyhydroxyalkanoates as value-added byproducts of a wastewater treatment plant, Processes 13 (2025), https://doi.org/10.3390/pr13020295.
[195] A. Ahmed, Y. Makkawi, The biorefinery paradigm: technologies, feedstocks, and retrofitting for future sustainable energy, Energies 18 (2025), https://doi.org/10.3390/en18225919.