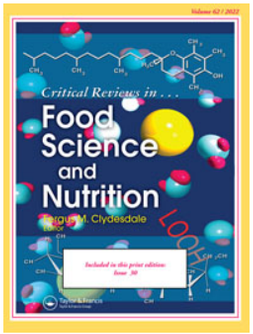

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REVIEW

Sources, purification, immobilization and industrial applications of microbial lipases: An overview

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ABSTRACT

Microbial lipase is looking for better attention with the fast growth of enzyme proficiency and other benefits like easy, cost-effective, and reliable manufacturing. Immobilized enzymes can be used repetitively and are incapable to catalyze the reactions in the system continuously. Hydrophobic supports are utilized to immobilize enzymes when the ionic strength is low. This approach allows for the immobilization, purification, stability, and hyperactivation of lipases in a single step. The diffusion of the substrate is more advantageous on hydrophobic supports than on hydrophilic supports in the carrier. These approaches are critical to the immobilization performance of the enzyme. For enzyme immobilization, synthesis provides a higher pH value as well as greater heat stability. Using a mixture of immobilization methods, the binding force between enzymes and the support rises, reducing enzyme leakage. Lipase adsorption produces interfacial activation when it is immobilized on hydrophobic support. As a result, in the immobilization process, this procedure is primarily used for a variety of industrial applications. Microbial sources, immobilization techniques, and industrial applications in the fields of food, flavor, detergent, paper and pulp, pharmaceuticals, biodiesel, derivatives of esters and amino groups, agrochemicals, biosensor applications, cosmetics, perfumery, and bioremediation are all discussed in this review.

KEYWORDS

Enzyme-modified cheese; food industry; immobilization techniques; Microbial lipases; pharmaceuticals; phospholipase

Introduction

Lipases (triacylglycerol acylhydrolase, EC 3.1.1.3) are hydrolases that operate on carboxylic ester linkages (Kurtovic et al. 2009). Triglycerides are hydrolyzed naturally by lipases into mono and di-glycerides, fatty acids, and glycerol (Houde, Kademi, and Leblanc 2004; Javed et al. 2018). It is known as a particular enzyme since it has a unique method of action called interfacial activation (Gonçalves, Silva, and Guidini 2019). In homogeneous conditions, most lipase molecules contain a polypeptide chain termed a lid covering their active center, which can separate it from the reaction medium (Yaacob et al. 2019). Fixing a new structure in the presence of a hydrophobic surface where the enzyme becomes adsorbed and the active center is fully exposed, allowing lipases to hydrolyze oil drops (Palomo et al. 2004). This approach was utilized to selectively immobilize lipases by their open forms on a variety of hydrophobic supports (Sakai et al. 2010; Bezerra et al. 2017; Klein et al. 1997). Lipase molecules are stabilized in their open and active forms on the support surface due to high adsorption (Alnoch et al. 2018). The anhydrous form of organic media used to accelerate biotransformation with methacrylate-like hydrophobic supports and lipase derivatives are solvents and solvent-free systems (Fernandez-Lorente, Rocha-Martín, and Guisan 2020).

The solubility of water enzymes was explained using immobilized techniques (Sheldon 2007; Zdarta et al. 2018). An encapsulation step is a valuable approach to improving enzyme functionality on an industrial scale. Immobilization occurs after other enzyme properties such as immobility, activity, specificity, selectivity, and resilience to inhibitors and chemical reagents have been developed (Bilal et al. 2019; Fernandez-Lafuente 2009; Garcia-Galan et al. 2011; Iyer and Ananthanarayan 2008; Mateo et al. 2007; Rodrigues and Ayub 2011). Immobilization can be used in conjunction with any other technique for enhancing enzyme characteristics. As a result, enzyme immobilization has become a prominent step in the creation of a commercial enzyme biocatalyst. If appropriately engineered, it can be used to purify the enzyme throughout the immobilization process (Barbosa et al. 2011). Physical and chemical techniques are preferred in industries for enzyme immobilization because they are easy and cost-effective (Mohamed et al. 2021). It's critical to understand the drawbacks and benefits of immobilized enzymes (Table 1). Amylases and proteases are drastically reduced when immobilized due to diffusion constraints, and immobilized enzymes are less economical

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Table 1. Advantages and disadvantages of immobilized enzymes in industrial processes.

Advantages	Disadvantages
• Easy separation of biocatalyst • In downstream processing costs reduced • Several uses of biocatalyst (reutilizing) • Toward organic solvents and higher temperatures, it shows better stability • Without the need of membrane to isolate enzyme from product use of the fixed bed or batch reactors • With other enzymes co-immobilization is possible	• Compared to the native enzyme it has a slow enzyme activity • For carriers and immobilization, additional costs required • Compared to native enzymes reaction rates slow • Subject to fouling • Exhausted immobilized enzyme incinerated (disposed)

due to the quick kinetics of native enzymes (Basso and Serban 2019).

The presence of lipase observed in bacterial species is *Bacillus prodigiosus, B. pyocyaneus, B. fluorescens*, and *Staphylococcus pyogenesaucreus* (Salwoom et al. 2019; Mobarak-Qamsari, Kasra-Kermanshahi, and Moosavi-Nejad 2011), and the other significant species are *Bacillus, Pseudomonas, Burkholderia*; and fungal species are *Aspergillus, Penicillium, Rhizopus, Candida*, *Hypocrea pseudokoningii* and the species of yeasts are Zygosaccharomyces, *Pichia, Lachancea, Kluyveromyces, Saccharomyces, Candida*, and *Torulaspora,* detected to harvest lipase enzyme (Pereira et al. 2014; Bora, Gohain, and Das 2013; Alcazar-Valle et al. 2019).

Microbial lipases are highly favored in the enzyme market, accounting for about 90% of the global lipase market from bacteria and fungi (Chandra et al. 2020b). With the aggregate usage of lipase in several feeds for livestock in the worldwide market, the global lipase market was valued at USD 585.56 million in 2020 and is expected to reach USD 961.85 million by 2028, rising at a CAGR of 6.4 percent from 2021 to 2028. In the animal feed and dairy industries, there is an increasing demand for lipases to improve the value of meat and treated dairy commodities (Guerrand 2017). Microbial lipases are more reliable than vegetable and animal enzymes, and because of their high consistency, optimization, and process modification, they can be easily and cost-effectively produced through fermentation procedures with less time and space requirements (Andualema and Gessesse 2012; Raveendran et al. 2018; Rosenthal and Lütz 2018). It aids in the refinement of food value and improves the taste, texture, shelf life, and smoothness of some products in the food sector as a nutrient (Barrett, Beaulieu, and Shewfelt 2010). Lipases have been employed on a huge scale to improve the health fitness of numerous animal feeds on the global market, and are mostly used in animal feeds, bakery, dairy, and confectionery sectors. Because of their numerous applications in a wide range of food processing from microorganisms, significant advancement is envisaged soon (Amit et al. 2017). The advantages of microbial lipases over animal and plant lipases are also supporting market growth (Bilal et al. 2021). Other actions covered involve interesterification, esterification, aminolysis, and alcoholysis, as well as relevant industries (Lee et al. 2001; Lima et al. 2017; Ismail and Baek 2020). Lipase produces esters from glycerol and long-chain fatty acids in a non-aqueous media. Flavor enhancement lipases are considered a potential biocatalyst in a variety of biotechnological processes, particularly in dairy products such as cheese recovery (Dandavate, Keharia, and Madamwar 2011). Increased consumption of enzyme-modified cheese (EMC) and enzyme-modified dairy ingredients (EMDI) as a result of a growing perception of animal strength and value has boosted the lipase market tremendously (Carneiro et al. 2020). In oil chemistry, bio-detergents are manufactured to remove harsh chlorine bleach from fresh diminishing sewage and industrial pollution (Fukunaga et al. 1998). Ibuprofen, naproxen, pregabalin, and ketoprofen, as well as (S)-Prosimpal, (S)-Piperoxan, (S, S)-Dibozane, and (S)-Doxazosin, are all manufactured in the pharmaceutical industry. RAC -ibuprofen (R, S)-2-(isobutylphenyl)-propionic acid) is a racemic mixture of ibuprofen (R, S)-2-(isobutylphenyl)-propionic acid (Adrio and Demain 2014; Sanchez and Demain 2011). The usage of recombinant DNA technology is a very appealing characteristic that can be exploited to overcome the economic constraints of industrial lipase use (Kanmani, Aravind, et al. 2015). Novozymes introduced Lipolase, the first lipase made with recombinant DNA technology, to the market in 1988. This lipase was developed in *Aspergillus oryzae* and came from *Thermomyces lanuginosus* (Gerits et al. 2014; Sarmah et al. 2018; Boel et al. 1988; Lin et al. 2016). Lipophilic extractives include alkanes, fatty alcohols, resin acids, fatty acids, conjugated sterols, certain terpenoids, waxes, and triglycerides, which are non-polar extractable fractions from wood and other lignocellulosic materials known as wood resin (Gutiérrez, Río, and Martínez 2009). Removing the esters from pulp lipase could be employed to improve manufacturing capacity and quality (Horchani et al., 2012).

Palatase from *Rhizomucor miehei* (Handayani et al. 2016), a commercial lipase articulated in *A. oryzae*, was another commercial lipase articulated in *A. oryzae* (Ansorena et al. 1998; Sankaran, Show, and Chang 2016). Because of their excellent wetting properties, wax esters are widely used in cosmetics, medicines, greases, and other biochemical products (Zalacain et al. 1997; Zhang, Aryee, and Simpson 2020). Immobilization, manufacturing, purification, categorization, and application of microbial lipases from a variety of sources were highlighted in this review. Immobilized enzymes include a variety of commercial lipases, as well as their immobilization methods.

Explanation of lipases

Lipases naturally hydrolyze mono, di, triglycerides, fatty acids, and glycerol into mono, di, triglycerides, fatty acids, and glycerol (Schomburg, Chang, and Schomburg 2014; Chahinian and Sarda 2009). Carboxylic esters linkages can

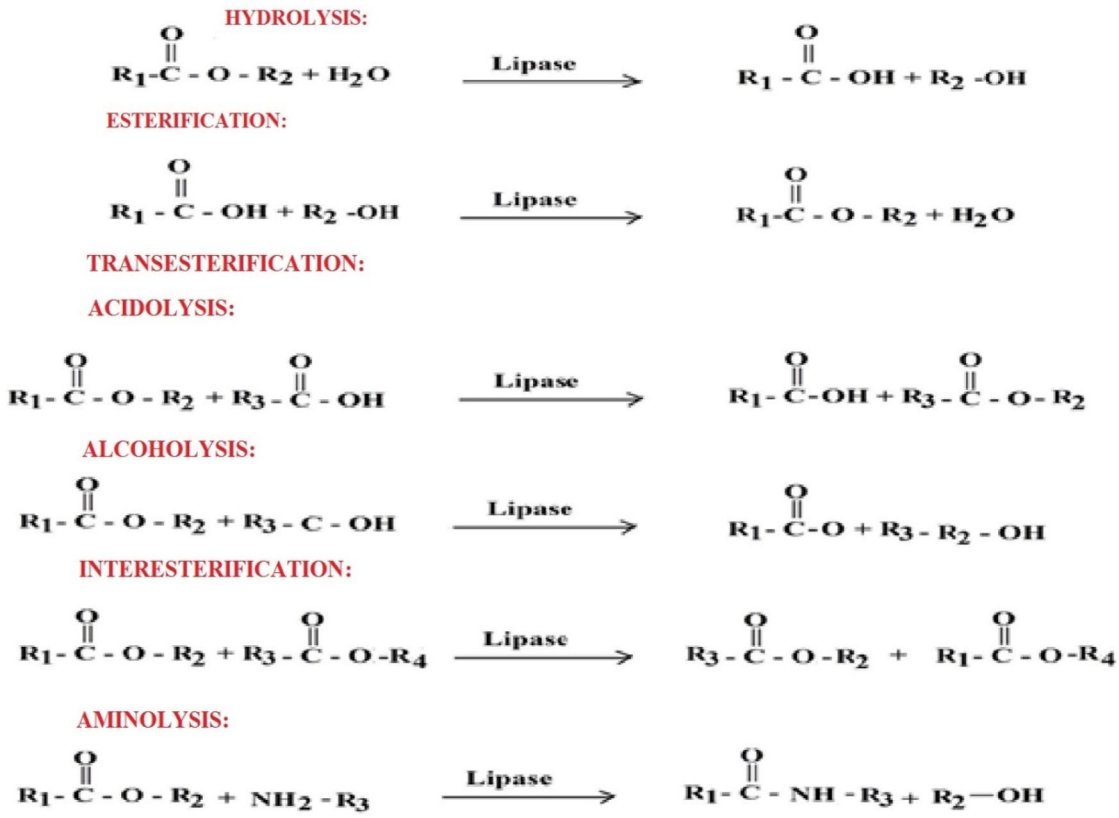

Figure 1. Reactions catalysis using lipase.

be degraded by enzymes other than lipases and esterases (Levisson, Oost, and Kengen 2009). For a long time, the distinction between lipases and esterases has been based on interfacial activation and the presence of a lid for the former enzyme (Schmid and Verger 1998; Brocca et al. 2003). Lipase activity rises rapidly in the presence of an interface, which is known as interfacial activation, whereas an amphiphilic surface loop known as cover shields the active site of lipase in solution and travels away with the interface when the two come into contact (Eggert et al. 2004). Despite the presence of a lid, *Pseudomonas aeruginosa*, *Candida anatarctica* B (Theil and Björkling 1993), and *Burkholderia glumae* lipases do not show interfacial activation (Li and Zhang 2016; Hotta et al. 2002; Kovacic et al. 2019). The presence of a lid and interfacial activation can simply be explained as a carboxylesterase that catalyzes the hydrolysis and production of long-chain acylglycerols and cannot be classified as a real lipase (Lopes et al. 2011; Chamorro et al. 1998).

Extracellular lipase

Extracellular lipases were created by microbes through solid-state or submerged fermentation (Sales et al. 2020). An enzyme's biocatalytic activity is frequently regained by raising the degree of purity in the fermentation process and then purifying it (Borkar et al. 2009). Extracellular lipase production is a complex process regulated by the origin and structure of the lipase (Palekar, Vasudevan, and Yan 2000; Doolittle and Péterfy 2010). Extracellular lipases should be cost-effective, rapid, simple, and efficient when used in large-scale production (Ventura and Coutinho 2016; Robinson 2015; Soleymani et al. 2017). In the vast majority of cases, extracellular immobilized lipases are commercially accessible (Ingenbosch et al. 2019; Robles-Medina et al. 2009). *Candida antarctica*, *Rhizomucor miehei*, and *Thermomyces lanuginosus* were used to identify and purify the immobilized lipases Lipozyme RM IM and Lipozyme TL IM, Novozyme 435 (Hernández-Martín and Otero 2008; de Souza et al. 2018; Bueso et al. 2015; Kobayashi 2011; López-Serrano et al. 2002; Wang et al. 2014). That is, the support may first immobilize through a one-point or multipoint interaction (Sunitha et al. 2007; Seitz 1974), and then, as with heterofunctional supports, it may continue to increase the number (or even the quality) of interactions, including new groups (Muralidhar et al. 2002; Ban et al. 2002).

Intracellular lipase. The utilization of entire cells as biocatalysts is an alternate way for resolving the high cost of extracellular lipase purification. Intracellular lipase refers to the use of lipase that has been liberated from cells (Schoemaker, Mink, and Wubbolts 2003; Adamczak and Bednarski 2004). Intracellular lipase is lipase that is secreted from the cell's outer surface. Because of certain supports, certain microorganisms can naturally immobilize castoff as a lipase source (Uthoff, Bröker, and Steinbüchel 2009). It eliminates the requirement for an

expensive purification step as well as the necessity for a lengthy immobilization technique, which is required with extracellular lipase (Ferreira-Dias et al. 2013; Adlercreutz 2013).

Reactions catalyzed by lipases

Lipase catalyzes the hydrolysis of carboxylate ester bonds at the organic-aqueous interface in the presence of excess water, releasing organic alcohols and free fatty acids (FFAs) (El Seoud, Baader, and Bastos 2016; Schoffelen and Hest 2013). The equilibrium was evaluated under the limitation of the reverse reaction using the water activity (aw) of the reaction mixture among the forward and reverse reactions (Lewis et al. 1965). Under low water activity, a variety of transesterification processes can be carried out (aw) (Bovara et al. 1993). An ester and alcohol (alcoholysis), an ester and acid (acidolysis), an ester and amine (aminolysis), or an ester and two esters (interesterification) undergo transesterification (Figure 1) (Santilli et al. 1987).

Lipases have numerous types of selectivity for their substrates, including chemo-specificity: which shows both fatty acid (FA) and lipid-class specificity when utilizing lipases (Raclot, Holm, and Langin 2001; Albayati et al. 2020). FA specificity is connected to the length or degree of unsaturation that declines within specific kinds of FFAs (Lennen and Pfleger 2012; Barros, Fleuri, and Macedo 2010). For lipid-class specificity, lipases catalyze the hydrolysis of mono, di, and triacylglycerol; (ii) Regio-specificity: General lipases catalyze the entire hydrolysis of TAGs into glycerol and FFAs in an unplanned manner, producing DAGs and MAGs as intermediates (Chang and Lee 2021). Using 1, 3-specific lipases, the TAGs were hydrolyzed solely at the sn-1 and sn-3 locations, yielding FFAs, 1, 2-DAGs or 2, 3-DAGs, and 2-MAGs (Frayn et al. 2003; Ishchenko et al. 2017). Because 2, 3-DAGs, and 2-MAGs are highly unstable, they undergo acyl migration to create 1, 3-DAGs and 1-MAGs or 3-MAGs, respectively, (iii) Enantio-selectivity: Lipases can tell the difference between the two enantiomers in a racemic combination (Jaeger and Reetz 1998). This selectivity fluctuates with admiration for the substrate, as it is related to the nature of esters.

Production and purification of lipases

In this section, we discuss the current advancements in lipase manufacture through host strain and metabolic engineering processes, as well as the selectivity, stability, and broad substrate specificity of microbial lipases most commonly employed in industry (Anobom et al. 2014).

Host strain selection

The most capable option for lowering the price of lipases is to use heterologous technologies for the creation of functional lipases (Macrae 1983; Baneyx 1999). Several types of microorganisms have been industrialized for heterologous and homologous manifestation in adept lipase host strains since the ancient period summarized in the given Table 2 (Contesini et al. 2020).

Bacteria

E. coli is the most commonly used manifestation host for recombinant protein expression for a variety of reasons. For hereditary employment, *E. coli* is more adaptable, and for genetic employment, it has higher refurbishment proficiency and faster progress rates (Rosano and Ceccarelli 2014; Ozturkoglu-Budak et al. 2016). However, because of a lack of adequate folding machinery, the typical *E. coli* system leads to the intracellular formation of sluggish or intractable inclusion bodies (Blank et al. 2006). *Candida antarctica* (CalB) is well-known in the biocatalysis industry for producing Lipase B, an active version of the enzyme that can be expressed in *E. coli* by changing the reaction standard or lipase transformation (Paraskevopoulou and Falcone 2018; Kundys et al. 2018; Wu, Yang, and Ge 2017). Lipases fused can increase the solubility of proteins in *E. coli* containing a polycationic amino acid tag. Furthermore, certain types of lipases require the formation of specific disulfide bonds for helpful proteins to be transported (Liebeton, Zacharias, and Jaeger 2001; de Marco 2009). This issue can be solved by using a certain *E. coli* Origami (DE3) strain or co-expression of a Dsb-family protein, such as DsbA, which is involved in disulfide bond formation (Urban et al. 2001; Vieira Gomes et al. 2018).

Yeast

As expression systems for complicated proteins, yeasts have various advantages, including strong growth capabilities, sanctioning disulfide bond formation, informal genetic manipulation, and post-translational protein processing (Lobstein et al. 2012). Under the control of methanol-responsive alcohol oxidase promoters, *K. phaffii* has the capabilities to express and produce lipases. In the extracellular medium it secretes fewer quantities of proteins naturally, but using methanol as an inducer high level of recombinant lipase can be achieved. The lipase purification step is easier and cost-effective makes by these special features (Cregg et al. 2000). *Saccharomyces cerevisiae* has been designated as a nonpathogenic host for the synthesis of heterologous lipase for a limited time (Darvishi 2012). *Yarrowia lipolytica* effectively transformed Lipase 2 (LIP2) gene into *S. cerevisiae* using PEX11 promoter (Shockey et al. 2011), and Lip2 lipase (Lip2p) was active in growth augmentation (Liu et al. 2012).

However, the *S. cerevisiae* expression system sanctions elevated heterologous protein expression, and inherent manipulation has numerous weaknesses like poor plasmid strength, less release ability, striving in scale-up, and hyper-glycosylation. *Pichia pastoris* used as a host for the production lipases (Passolunghi et al. 2003). It presents some benefits, like a highly-regulated promoter of the alcohol oxidase (AOX) over other hosts (Zhang et al. 2010; Li et al. 2010). It can be grown up to exceedingly more mass of cells in eukaryotic minimal medium and proteasome secretion

Table 2. Heterologous and homologous production of lipase using the prokaryotic and eukaryotic system.

Enzyme source	Enzyme	Expression vector (Expression host)	Cloning vector (Recipient strain)	**Expression vector**	Remarks	References
Acinetobacter haemolyticus	Lipase KV1	*E. coli* BL21 (DE3)	pGEM-T Easy (*E. coli* JM109)	pET-30a (+)	Using Response Surface Methods (RSM) optimization of production conditions	Batumalaie et al. 2018; Fabiano Jares Contesini et al. 2020.
Bacillus amyloliquefaciens G7	Lipase BaG7Lip	*E. coli* BL21-Star (DE3)	p15TV-L (*E. coli* BL21-Star (DE3))	p15TV-L	Metagenomics libraries construction and using a boolean network analysis prediction of the best-producing conditions. Activity enhancement with acetone, glycerol, and K^+ ions	Khan et al. 2020.
Burkholderia contaminans LTEB11	Lipase LipBC (LipA) + foldase LifBC (LipB)	*E. coli* BL21 (DE3)	–	pET28a (+) and pT7-7 for LipA and LipB, respectively	Lipase and chaperone genes co-expressed. Specific activity of 1426 U/mg (tributyrin); > Over five reaction cycles of 6 h at 45 °C, 80% conversion of ethyl-oleate in n-hexane.	Alnoch et al. 2018.
Burkholderia stabilis FERMP-21014	Cholesterol esterase + foldase	*E. coli* BL21 (DE3), *E. coli* Rosetta (DE3) and *B. stabilis*	*E. coli* DH5α and JM109	pET26b(+), pET39b(+), pET40b(+), pBBR122	Promoters screening by RNA-Seq + co-expression of lipase and foldase + heterologous and homologous expression. Recombinant activity was 243 fold higher than the WT without oleic acid; *B. stabilis* system was more efficient to produce esterase.	Yoshida et al. 2019.
Burkholderia territorii GP3	Lipase LipBT and foldase LifBT	*E. coli* DH5α, *E. coli* DH10β, *E. coli* BL21 (DE3) pLysS, *E. coli* Origami B, E. coli Shuffle B, and *E* . *coli* SHuffle K	pGEM-T Easy (*E. coli* DH5α), pET15b (*E. coli* DH10β)	pGEM-T Easy and pET15b	Metagenomics for screening and Identification of lipolytic strains and meta genomic for screening and best expression system evaluation. Higher lipase activity in *E. coli* BL21 (DE3) pLysS (pET15b) (6.73 ± 0.24 U/mg); optimum substrate pNP-C10; activity enhancement in n-hexane, Triton X100, and Ca^{2+} and Mg^{2+} ions.	Putra et al. 2019.
Clostridium acetobutylicum (ATCC 824)	Lipase Ca-Est	*E. coli* BL21 (DE3)	pMCSG7 (*E. coli* DH5α)	pMCSG7	Rational design, docking analysis, site-directed mutagenesis. Activity enhancement with methanol; residues Ser89 and His224 are crucial for catalysis.	Nagaroor, V and Gummadi, 2020.
Candida antarctica	Lipase CALB	*Corynebacterium glutamicum* MB001	pEC-CALB and pEC-H36-CALB (*E. coli* DH5α)	pEC-CALB and pEC-H36-CALB	Using 10 mM MgSO4 31% activity was inhibited.	González et al. 2019
Drosophila melanogaster	Lipase Lip3	*E. coli* BL21 (DE3)	-	pETMCSIII	Directed evolution, error-prone PCR, construction of variant libraries. R7_59A mutant activity was higher than the WT toward tributyrin, glyceryl trioctanoate, coconut oil, glyceryl trioleate, and pNP (pNP-C3, pNP-C8, pNP-C16, pNP-C18) substrates; 57_59A activity enhancement of 228 fold compared to the WT using pNP-C8 substrate.	Alfaro-Chávez et al. 2019.
Geobacillus zalihae	Lipase HT1-5M	*E. coli* BL21 (DE3) pLysS	pUC57 and pGEX-4T1 (*E. coli* TOP10)	pLysS	Rational design, molecular dynamics (MD), site-directed mutagenesis. Activity enhancement with Ca^{2+} ions; more stable in DMSO, n-hexane, and n-heptane with Ca^{2+} ions	Ishak et al. 2019.
Proteus sp. NH 2-2	Lipase LipPN1	*E. coli* BL21 (DE3)	-	pET-28a (+)	Site-directed mutagenesis. Highest activity toward pNP-butyrate (pH 9.0, 40 °C); activity enhancement with acetone and ions Ca^{2+}, Mn^{2+} and Mg^{2+}; rLipPN1 and rLipPN1_C85S reached 1662 U/L and 1436 U/L, respectively; 91.5% of soybean oil was converted into biodiesel.	Shao et al. 2019.
Pseudomonas fluorescens AMS8	G55Y, T52Y and AMS8 recombinant lipases	*E. coli* BL21 (DE3)	-	pET32b	Rational design, site-directed mutagenesis. G55Y and T52Y lipases had lower affinity by pNP-palmitate, laurate and caprylate substrates compared to WT AMS8 lipase; efficiency of G55Y lipase was higher than T52Y for pNPL and pNPP substrates.	Yaacob et al. 2019.

(*Continued*)

Table 2. (Continued).

Enzyme source	Enzyme	Expression Vector (Expression Host)	Cloning Vector (Recipient Strain)	**Expression Vector**	Remarks	References
Pseudomonas sp. LSK25 E	Lipase LSK25	*E. coli* BL21 (DE3)	pGEMT Easy (-)	pET32b(+)	Activity enhancement with Ca2+ ions; activity boosted in toluene, xylene, n-hexane, n-heptane, and n-hexadecane; higher activities toward long-chain fatty acids contained in coconut oil and rice brain oil, and pNP-C12.	Salwoom et al. 2019.
Heterologous production of lipases using the eukaryotic system						
Marine *Streptomyces* sp. strain W007	Lipase MAS1	*K. phaffii* X-33	pPICZαA (*E. coli* DH5α)	pPICZαA	PDI co-expression gives 1.7 fold increases lipase expression. The highest lipase production was achieved at pH 6.0 and 24° C with an activity of 440 U/mL.	Lan et al. 2016.
Rhizopus chinensis	r27RCL	*K. phaffii* GS115	pPIC9K	pPIC9K	PDI co-expression gives 1 fold increase. The highest lipase activity reached 355 U/mL with one copy of PDI and five copies of r27RCL gene.	Sha et al. 2013.
Candida antarctica	CALB	*K. phaffii* X-33 and M12 (leu2)	*E. coli* Stellar™ and XL10-Gold®	pBluescript II SK pPGKΔ3PRO_ LIPB	Strain with three copies achieved 48.760 U/L enzyme yield, 2.3 fold higher than the one-copy strain.	Robert et al. 2019.
Candida antarctica	CALB	*K. phaffii* GS115	pPICZαB (E. coli TOP10F′)	pPICZαB	Combinatorial overexpression of Ydj1p-Ssa1p resulted in the highest fold increase, 2.5. Individual overexpression of Ydj1p, Ssa1p, and Sec63p increased CALB expression level by 1.6, 1.4, and 1.4 fold, respectively. Co-expression of Ydjlp-Sec63p Kar2p-Ssalp, Kar2p-Sec63p, resulted in 1.5, 1.5 and 1.5 fold increases, respectively. Kar2p-Ydj1p combination resulted in decreased CALB secretion.	Samuel et al. 2013.
Rhizopus oryzae	ROL	*K. phaffii* GS115	pPICZα and pAO815 (*E. coli* Top10 cells)	pPICZα-ROL	Strain with five copies resulted in 8 fold increase in ROL expression. Co-expression of both genes Ubc1 and Hrd1 resulted in.54.2% higher ROL activity, 4750 U/mL.	Jiao et al. 2018
Fusarium solani	FSL	*K. phaffii* X-33	pPICZαA and pGAPZαA (*E. coli* DH5α)	pPICZαA-FSL and pGAPZαA-FSL	Strain expressing pGAPZαA-FSL produced the highest specific lipase activity, 18.81 ± 1.98 U/mg, in 3 days of cultivation time. Lipase activity was enhanced by Mn2+, Ba2+, Li+, Ca2+, Ni2+, CHAPS, and Triton X-100 but was inhibited by Hg2+, Ag+, and SDS.	Wongwatanapaiboon et al. 2016.
Rhizopus oryzae	ROL	*K. phaffii* X-33	–	pPICZFLDαROL	pFLD gave a 1.9 fold increase compared to pAOX1. The best ROL production strategy with the PFLD-based system is a fed-batch induction phase with a constant sorbitol excess.	Resina et al. 2005.
Candida antarctica	CALB	*K. phaffi* GS115 and *Y. lipolytica* RIY368	*E. coli*	*K. phaffii* (pIB4, promoter pAOX1, HIS4 marker) and *Y. lipolytica* (JMP4266, promoter pEYK1-A3B, URA3 marker)	After 72 h cultivation, the lipase activity was 5540 U/mg dcw for *Y. lipolytica* strain RIY368 and *K* . *phaffii* strain RIY311 1066 U/mg dcw for *Y. lipolytica*, during the growth phase the lipase activity increased, whereas during growth and stationary phase in both conditions for *P. pastoris* it increased	Theron et al. 2020
A. niger	Lipase lip	*T. reesei* Tu6 strain	pBluescript II SK(+) and pMD18-T Simple (*E. coli* DH5α)	pSKpyr4 and pSK-lip	Higher levels of lipase all cph1 gene silencing transformants expressed and less cbh 1 transcript than the reference strain, approximately lower than 2%. The RNAi mediated gene silencing of cbh 1 did not negatively affect the lipase transcript abundance.	Qin et al. 2012.
Thermomyces dupontii	TDL	*K. phaffii* X33	pPICZαA (E. coli Top 10)	PICZαA–tdl-opt	The highest TDL activity was achieved with pFLD1 (27.076 U/mL), of 15 methanol-inducible promoters, whereas of nine constitutive promoters, pGCW14 gave the highest activity (17.353 U/mL).	Wang et al. 2019.
Meyerozyma guilliermondii strain SMB	L2 lipase	*Komagataella phaffii*	*Bacillus* sp. (pFLDhα)	pFLDhα	To express L2 lipase under the regulation of P_{FLD1}, A new host-vector system was established as a platform.	Salleh et al. 2020.

at low levels and post-translational alterations of proteins. It can also secrete heterologous protein hosts with ease (Tokmakov et al. 2012; Ahmad et al. 2014). *Rhizophus* sp. lipase, for example, is not expressed in *E. coli* due to a lack of necessary proteases to process fungal maturation signals. However, it can be successfully expressed in the *P. pastoris* host (Karbalaei, Rezaee, and Farsiani 2020; Contesini et al. 2020). *P. pastoris* ability to release heterologous target proteins extracellularly with less contaminating proteins is often employed to reduce process costs (Löbs, Schwartz, and Wheeldon 2017). *P. pastoris* has been identified as the most capable host for heterologous lipase construction among several yeast strains, particularly from eukaryotic sources (Karbalaei, Rezaee, and Farsiani 2020). Non-conventional yeasts such as *Hansenula polymorpha* and *Yarrowia lipolytica* have been proposed (Abdala et al. 2017; Zhou et al. 2019). These strains operate differently depending on the type of heterologous protein used. Due to its increased lipase production (Madzak, Gaillardin, and Beckerich 2004; Gasmi et al. 2011) and adaptation for CalB synthesis between these non-conventional strains, *Y. lipolytica* appears to be an added potential replacement host (Valero 2012).

Fungi

The big lipase-producing bases include *Mucor, Rhizopus, Geotrichum, Rhizomucor, Aspergillus,* and *Penicillium* (Ayinla et al. 2021). As with yeasts and bacteria, filamentous fungus hosts are assessed as a supplement. Extracellular protein secretion in heterologous hosts filamentous fungi benefit from a variety of factors, including increased plasmid copy number, plasmid stability, and capability (Contesini et al. 2010; Rantasalo et al. 2019). Business bids for lipase production between rehabilitated fungal species frequently involve *Aspergillus* and *Trichoderma* sp. (Tamalampudi et al. 2007; Hwang et al. 2004). Like most alert filamentous fungi, *Aspergillus oryzae* is the host; CalB with higher esterification activity has been heterologously fashioned by *Aspergillus oryzae* and immobilized for whole-cell biocatalyst for enzymatic biofuel generation (Adachi et al. 2013; Hwang et al. 2014; Budhwani et al. 2019). In recent years, *Trichoderma reesei* has been considered as an alternative host for recombinant protein production using the cbh1 promoter and has been measured for recombinant lipase production (Qin et al. 2018).

Host strain engineering

Various methods are used to increase the production of massive lipase, which is necessary for the commercial application of lipases. To maximize lipase productivity, we investigate a variety of techniques and metabolic engineering performances (Ward 2012).

Genetic manipulation of host strains

Microorganisms produce a wide range of extracellular lipases, which are then processed into commercial lipases (Fickers, Destain, and Thonart 2009; Gupta, Gupta, and Rathi 2004). Using the fed-batch fermentation procedure, a significant increase in lipase production has been achieved. Lip2 produced a higher yield of lipase when two distinct orders of *Y. lipolytica* mutant-strain LgX64.81 were used (Yu, Wen, and Tan 2010; Batumalaie et al. 2018). The most promising techniques to achieving large amounts of purified lipases to fulfill the standard of quantity and manipulation of industrial procedures are replicating and recombinant lipase gene expression (Farrokh, Yakhchali, and Karkhane 2014; Krzeslak et al. 2008). In most cases, the castoff technique is promoter optimization, which significantly enhances lipase manufacture (Meunchan et al. 2015). Strong constitutive marketers like XPR2, TEF, and RPS7 (Trassaert et al. 2017) and inducible supporters like ICL1, POT1, and POX2 (Park, Do, and Jung 2013) have been promoted for *Y. lipolytica* lipase yield (Liu et al. 2013). *Y. lipolytica* was not measured as a perfect mass because it lacked a "perfect" inducible promoter. Heterologous expression of the protein is identified in the *E. coli* system by the intracellular gathering of sluggish or convoluted inclusion forms (Hellwig et al. 2004; Xu, Lewis, and Chou 2008). *Pseudozyma antarctica* lipase B was co-expressed in *E. coli* with several periplasmic folding factors, including DegP, FkpA, DsbA, and DsbC (Narayanan, Khan, and Chou 2010). PalB (inclusion bodies) that are uneven and sluggish can help in the presence of these folding features (Ujiie, Nakano, and Iwasaki 2016; Dugourd et al. 1999). As a result, both the cytoplasm and periplasmic material improved significantly in the functional PalB expression. The ABC transporter protein is naturally employed for a wide range of substrate transformations, including ions, carbohydrates, and amino acids (García, Hoyos, and Hernáiz 2018; Navarre and Schneewind 1999; Van Geest and Lolkema 2000).

An inner membrane protein with trans membrane segments 6-8 and a C-terminal ATPase domain is composed of an N-terminal membrane area (Ahn, Pan, and Rhee 1999; Jaenicke and Böhm 1998). TliD, TliE, and TliF involving ABC transporters have been removed from *E. coli* to speed up the release of a thermostable lipase (TliA) (White et al. 2008). By co-expressing TliA with altered TliD, the secretion levels of TliA lipase were increased thrice while the expression level of transporter protein remained almost unaffected, indicating that designed transporter proteins can speed up the secretion of lipases (Baek et al. 2010; Eom et al. 2005). The process of determining the target proteins related to an attaching theme on the outside of many host cells is known as cell surface demonstration (Xu and Lee 1999). *E. coli* OmpC created a cell-surface display system as a fastening idea to boost the expression of *P. fluorescens* SIKW1 lipase TliA (Choi and Lee 2004). Lipase cell surface display appears to stress the cell due to its added heterologous protein "load," however in prokaryotic and eukaryotic host cells; this approach can greatly enhance protein production (Meadows, Kang, and Lee 2018; Hwang et al. 2014).

Using metabolic engineering lipase production improvement

Metabolic manufacturing has played a significant role in boosting biofuels production (Stephanopoulos 2007; Saeui et al. 2015). Beneficial protein output has also been increased through cultivation. Though metabolic manufacturing has been compared to lipase production (Son et al. 2012), it has been comparatively threatened in lipase production. In recent work for extracellular lipase yield, the metabolic manufacturing of *P. fluorescens* has been mentioned as an exception. This keeps a secretion system running, allowing a thermostable lipase enzyme to be excreted (Kanimozhi and Perinbam 2015). The recombinant protein degradation produced varied depending on the culture media types and air circulation, consuming the natural lipase (TilA) and protease genes SIK W1 of *P. fluorescens* using the directed gene knockout strategy. The deletion deformed *P. fluorescens* hidden recombinant lipase (TilA) at high levels in a blending usage without degradation regardless of growing conditions (Saxena et al. 2003). To investigate the co-production of D-psicose and lipase, *B. subtilis* and *E. coli* were created utilizing a recombinant co-culture method.

Purifications of lipase

Several novel purification approaches are available to acquire lipase homogeneity from a wide number of microorganisms (bacteria, fungus). Purification of lipases excavated on the clarity sought for foodstuff usage usually involves many processes. The fermentation approach is commonly employed for the removal of extracellular microbial lipases from the broth culture. The traditional purification procedures are ultrafiltration, precipitation, and affinity chromatography. However, several of these procedures are difficult, time-consuming, and costly.

Novel purification apparatuses like (i) membrane separation processes, (ii) immunopurification, (iii) hydrophobic collaboration chromatography by epoxy-activated spacer arm as a ligand and polyethylene glycol immobilized on sepharose, (iv) polyvinyl alcohol polymers as column chromatography stationary phases, and (v) aqueous two-phase systems are employed commonly (Gopinath et al. 2003). Hydrophobic interaction chromatography is commonly used for enzyme recovery and fold purification. An acid-resistant lipase was isolated from crude commercial preparations utilizing *A. niger* and size exclusion on Bio-gel-p-100 and ion exchange on Mono-Q (Oliveira de Medeiros, Burkert, and Kalil 2012; Sugo and Okuyama 2018). Lipase from *R. japonicus* NR400 was isolated to homogeneity using hydroxyapatite, octylsepharose, and sephacryl S-20037 chromatography (Niu, Wang, and Tseng 2006; Kojima and Shimizu 2003). For lipases purification from various sources, several distillation procedures are listed in Table 3.

Properties and characteristics of lipases

Galvão et al. (2018) identified lipases as one of the most important biocatalysts with established potential for contributing to bio-industry through lipid technology. They've been employed both in-situ and ex-situ in lipid metabolism and a variety of industrial applications (Delorme et al. 2011). Serine, aspartate, glutamate, and histidine make up the active site of lipases (Pinheiro et al. 2018), Lipases can be found in two distinct conformations, open (active) and closed (inactive), as a result of their interfacial activation (inactive). Lipases in their open state are said to be more stable than those in their closed state (Willems et al. 2017). In the presence of hydrophobic surfaces, the open form of lipases usually occurs with the movement of the lid, boosting enzyme activity (Stauch, Fisher, and Cianci 2015). The ultimate features of lipases, such as specificity and selectivity, are not considerably affected by this movement (Dobreva, Zhekova, and Dobrev 2019). Genetic manipulation or physio-chemical changes can easily change these traits (including immobilization).

Lipases are monomeric proteins with a molecular weight of 19-60 kDa (Sharma, Chisti, and Banerjee 2001). The physical features of lipases are primarily determined by factors such as the position of fatty acids in the glycerol strength, the length of the fatty acid chain, and the degree of unsaturation (Rengachari et al. 2012; Adlercreutz 2013). These characteristics have an impact on a triglyceride's nutritional and sensory value. Numerous lipases are active in organic solvents and catalyze esterification and a variety of other useful reactions (Urban et al. 2001). The amount of ammonium sulfate solution cast off during the refinement process determines the rise in lipase activity (Svendsen et al. 1997; Raftari et al. 2012; Borkar et al. 2009). *C. rugosa* lipases for butanol, pentanol, hexanol, propionic acid, and butyric acid, *M. miehei* and *R. arrhizus* lipases for long-chain acids and acetates, and *M. miehei* and *R. arrhizus* lipases for long-chain acids and acetates exposed major marketable lipases (Silva and Jesus 2003; Abramić et al. 1999; Paluzar, Tuncay, and Aydogdu 2021). Lipase activity is affected by pH, and some lipases are stable across a wide range of pH standards (Bakir and Metin 2016). Most lipases are stable at neutral pH, but others can withstand pH values as high as 4.0 and 8.0 (Hasan, Shah, and Hameed 2009). At acidic pH, *Chromobacterium viscosum, A. niger*, and *Rhizophus* species generated extracellular lipases (Woodcock et al. 2008). *P. nitroaeducens* alkaline lipase was isolated and found to be active at pH 11.0 (Rajendran, Palanisamy, and Thangavelu 2009). Lipases are capable of reversing the reaction that leads to esterification and interesterification under some uncertain settings, such as when water lipases are absent (Alvarez and Stella 1989; Akimoto et al. 2003; Nicholson and Marangoni 2021). Divalent cations, such as calcium, excite activity for the production of lipase action, and cofactors are usually not required (Lu et al. 2013; Falcocchio et al. 2006; El Khattabi et al. 2003). Lipase activity was strongly decreased by Co^{2+}, Ni^{2+}, Hg^{2+}, and Sn^{2+}, while Zn^{2+}, Mg^{2+}, EDTA, and SDS were only mildly inhibited (Winkler and Stuckmann 1979; Carrie, Delaquis, and Mazza 1999).

When *S. marcescens* SM-6 was exposed to glycogen, hyaluronate, pectin B, or gum Arabic, lipase growth was considerably improved, and spontaneous and cyclic AMP

Table 3. Purification methods for lipase enzymes.

Microorganism	Purification Method	References
***Pseudomonas* spp.**		
P. fluorescens HU380	Superdex – 200 HR FPLC, Phenyl-toyoperal fractionation (batch-wise), DEAE– sepharose column chromatography	(Nakai et al. 1968)
Pseudomonas sp.	Superose 12B chromatography, Biogel P-10 chromatography	(Lund and Granum 1996; Hiol et al. 2000)
P. fluorescens strain 2 D	Hydrophobic interaction chromatography, Ammonium sulfate precipitation	(Woods and Que 1987)
P. aeruginosa	Hydroxyapatite column chromatography, Ammonium sulfate precipitation	(Gilbert, Cornish, and Jones 1991)
P. aeruginosa EF2	Gel filtration (superose) FPLC, Ultrafiltration, Anion exchange chromatography (Mono Q)	(Ndaw et al. 2002)
***Bacillus* spp.**		
Bacillus sp.	Ammonium sulfate, Acrinol treatment, DEAE-Sephadex A-50, Toyopearl HW-55F, Butyl toyoperal 650 M.	(Vale et al. 1986)
Bacillus sp.	Acetone fractionation, Two acetone precipitations, Octyl-sepharose CL-4B, Q Sepharose, Sepharose-12.	(Li and Zong 2010)
B. thermoatenulatus	Calcium soap, Hexane extraction, Methanol precipitation, Q-sepharose (ion exchange).	(Sarkar et al. 2012)
***Staphylococcus* sp.**		
S. aureus	Hydrophobic chromatography on phenyl-Sepharose CL-4B	(Sato et al. 1999)
S. hyicus	Sephadex G-100/G-25 column	Taipa, Aires-Barros, and Cabral 1992)
***Rhizopus* spp.**		
R. japonicus KY 521	Sephadex G-100 gel filtration	(Farag and Hassan 2004)
R. oryzae	Acetone precipitation (80%), Sephadex G-100.	(Nascimento et al. 2012)
R. arrhizus	Ammonium sulfate fractionation and Sephadex G-100 gel filtration.	(Tahoun and Ali 1986)
R. delemar	Ultrogel and Sephadex G-150 columns	(Palissa et al. 1989)
***Penicillium* spp.**		
P. chrysogenum	Ultrafiltration, Phenyl-sepharose, Mono Q HR5/5, and PD-10 column on Sephadex G-25.	(Krieger et al. 1997)
P. citrinum	Extraction and back-extraction using AOT reversed micelles in isooctane and phenyl- superose column.	(Kimiyasu, Akiba, and Yamaguchi 1988)
Penicillium cyclopium M1	Ammonium sulfate, DEAE-cellulose, DEAE-Sepharose, hexyl-Sepharose, and hydroxyapatite chromatography and gel Filtration on Cellulofine GC-700.	(De Jong et al. 1992)
Penicillium simplicissimum	Phenyl-Sepharose column.	(Isobe et al. 1992)
Penicillium camembertii U-150	Ammonium sulfate fractionation, Aminooctyl-Sepharose, Hydroxyapatite	(Gargova et al. 2006)
***Aspergillus* sp.**		
Aspergillus niger	Hydrophobic interaction (butyl Toyopearl 650 M), Gel filtration (Sephadex G-75), Anion-exchange chromatography (DEAE-Sepharose CL-6B)	(Nagaoka and Yamada 1973)
***Mucor* lipases sp.**		
Mucor lipolyticus Aac-0102	CM-Sephadex column chromatography	(Whitaker 1963)
Mucor javanicus	Gel filtration on Sephadex G-200	(Ota and Yamada 1966)
Yeast Lipase		
Candida paralipolytica	M-Sephadex C-50 and DEAE-Sephadex C-50.	(Maruyama et al. 2000)

production was stimulated (Kambourova et al. 2003; Krebs et al. 2014). At 40-60 °C, thermophilic bacteriological lipases from Icelandic hot springs performed better. In the temperature range of 30-35 °C, Epipactis gigantean has a strong lipase activity (Augustyniak et al. 2012; Royter et al. 2009; Rogalska et al. 1993). Lipases can be divided into two groups based on the region-specificity they show when using an acyl glycerol substrate (Wang et al. 2017). Lipases in the first category have no specificity for regions and release fatty acids from all three places of glycerols (Stadler et al. 1995; Gonzalez-Baró, Lewin, and Coleman 2007). The second assemblies of lipases issue fatty acids regio-specifically from the outer 1 and 3 locations of acyl-glycerols (Borrelli and Trono 2015). These lipases hydrolyze triacylglycerol to produce free fatty acids 2-monoacylglycerol and 1, 2-(2, 3)-diacylglycerol (Dinanta et al. 2019; Brindley 1991). *A. arrhizus, R. delemar*, *C. cylindracea*, and *P. aeruginosa* have all demonstrated fractional stereo-specificity in the hydrolysis of triacylglycerols (Macrae and Hammond 1985). These enzymes are employed to sequester optically pure esters and alcohols that are left behind (De Maria et al. 2007; Stehr et al. 2003; Balcão, Paiva, and Malcata 1996).

Selection of supports

Immobilizations and support strategies have been established to increase enzyme activity (Tischer and Wedekind 1999). In the realm of hydrodynamic circumstances, the selection of support material can be a difficult subject depending on reaction media, settings, enzyme types, and safety policies (Spahn and Minteer 2008; Boller, Meier, and Menzler 2002). Physical and chemical variables such as pore size, hydrophilic/hydrophobic balance, and surface chemistry parameters provide diversity for enzyme attachment (Longo and Combes 1997). These changes in morphological and physical parameters can alter enzyme immobilization and catalytic activity (Ittrat et al. 2014). Organic and inorganic supports can be classed as organic or inorganic, and natural and manufactured polymers can be subdivided based on their chemical makeup (Zdarta et al. 2018).

For enzyme immobilization, several supports have been reported such as natural and synthetic polymers (Bezerra et al. 2015), gels (Meunier and Legge 2013), inorganic materials (Zucca and Sanjust 2014), nanomaterials like glass beads, alginate beads (Datta, Christena, and Rajaram 2013), activated carbons (Cho and Bailey 1977), alumina

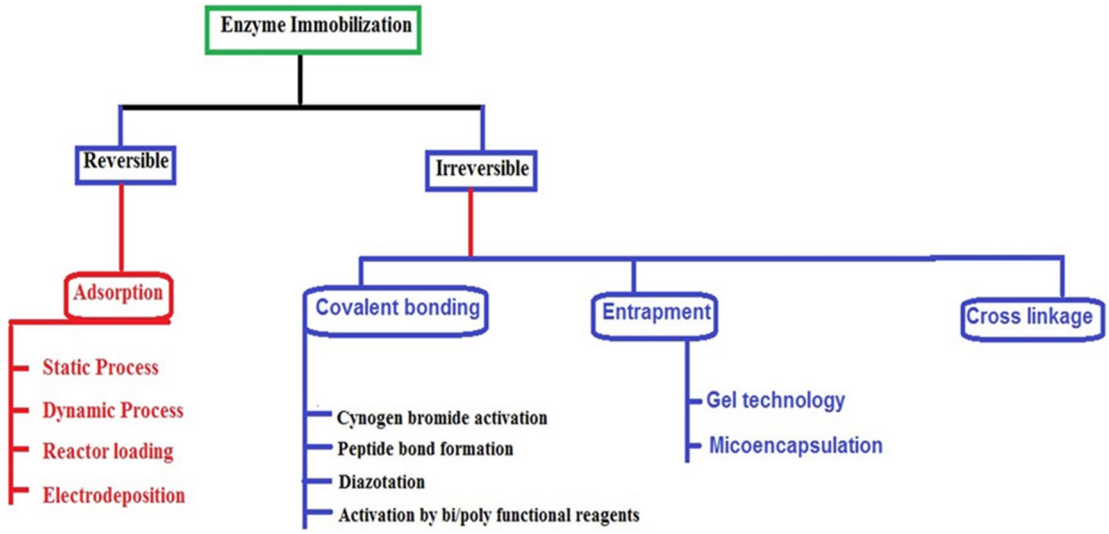

Figure 2. Categorizations and representation of different enzyme immobilization procedures.

(Sigurdardóttir et al. 2018), silica (Zdarta et al. 2018), celite (Sağiroğlu, Kilinç, and Elefoncu 2004), ceramic (Kujawa et al. 2021), metal oxides (Zucca, Fernandez-Lafuente, and Sanjust 2016), modified sepharose (Mohamed et al. 2021), or treated porous glass which is an organic material (Lee, Lin, and Mou 2009) and certain polymers are the most commonly used supports. One of the desired qualities of support matrix is mesoporous material, which has a higher number of pores and surface areas, resulting in increased enzyme loading per unit mass (Nematian et al. 2020). A technical process is used to fix enzymes to or within solid supports, resulting in a heterogeneous immobilized enzyme system (Krajewska 2004). The enzymes' structure is stabilized by solid support systems, and their activity is maintained (DiCosimo et al. 2013).

Strategies for immobilization

Enzyme immobilization is an efficient approach for improving the stability and reusability of enzymes even under adverse environmental conditions such as pH, temperature, and organic solvents, making enzymes more inexpensive and feasible in industrial production (Guzik, Hupert-Kocurek, and Wojcieszyńska 2014; de Souza et al. 2020). Because the elimination of lipases can increase the enzyme's selectivity, repeatability, substrate range, and separation from the raw materials, immobilization of the lipase enzyme is critical for its economic application (Chapman, Ismail, and Dinu 2018). Immobilization of lipase has several advantages, including better stability, lipase enzyme ejaculation halt, and greater lipase enzyme interaction with raw materials (Muniandy et al. 2019; Gonçalves, Silva, and Guidini 2019). Lipase enzyme contains a small number of lysine residues, making immobilization challenging. The lysine structure's functional groups play a significant role in matrix binding (Rafiee and Rezaee 2021). Its good solid support for lipase immobilization since it has available hydroxyl (-OH) and amine ($-NH_2$) functional groups: Enzyme immobilization is a straightforward and repeatable procedure that does not require specialized equipment (Zaitsev et al. 2019; Sharma et al. 2021).

Techniques for enzyme immobilization

Physical adsorption, ionic bonding, and covalent bonds are all maintained by enzymes (Casas-Godoy et al. 2012). Physical methods (reversible) and chemical methods (irreversible) are two types of enzyme immobilization methods (Pinheiro et al. 2019). Enzymes can be removed from the supporting material using reversible immobilization under mild circumstances. When enzymes adhere to supporting materials in irreversible immobilization, they are unable to be withdrawn without compromising the support or the enzyme's biological activity (Moreno-Pérez, Guisan, and Fernandez-Lorente 2014; Singh and Mukhopadhyay 2012; Xavier Malcata et al. 1990; Mead, Irvine, and Ramji 2002). Adsorption is a physical mechanism while cross-linking, covalent bonding processes, and entrapment is chemical methods (Harini et al. 2021; Liang et al. 2020; Soares et al. 1999; Gupta, Gupta, and Rathi 2004). In enzyme immobilization processes, a combination of these strategies has recently been applied (Liu, Schmid, and Rusnak 2006; Larios et al. 2004).

The kinetic characteristics of immobilized lipase improved significantly in terms of activity, specific activity, Km and Vmax, optimal pH, and temperature (Becker et al. 1997; Pereira et al. 2001; Gawas, Jadhav, and Rathod 2016). To examine the impact of immobilization on the enzyme's catalytic effectiveness, two kinetic parameters, the Michaelis constant Km and the maximal reaction velocity Vmax, are determined for an immobilized enzyme when compared to its non-immobilized counterpart (Neira and Herr 2017; Cooney 2017). Mechanical strength, chemical resistance, and microbial breakdown are all requirements for carrier architectures. On the other hand, on the surface of carriers, a large number of reactive groups and a hydrophilic chain were

required (Salvi, Kamble, and Yadav 2018). Other (desired or undesirable) events may emerge after immobilization, but the researcher must recognize them and devise instruments to regulate them. The diverse enzyme immobilization approaches and their categorizations are shown in Figure 2.

Physical methods or reversible

Adsorption

Organic polymers, charcoal, glass, mineral salts, metal oxides, or silica gel materials were adsorbed by enzymes. Nelson was the first to use this strategy, although the mechanics involved in adsorption aren't fully understood (Samiey, Cheng, and Wu 2014; Hartmann and Kostrov 2013; Sugahara and Varéa 2014). This method of regulating enzymes has the advantage of being both cost-effective and chemically modest; additionally, no chemicals are required, and the approach only involves a few triggering stages (Nguyen and Kim 2017). Furthermore, when compared to biochemical approaches for immobilizing enzymes, the process of immobilization enzyme is a lesser amount expected for a period to be denatured (Sheldon and van Pelt 2013). To express the immobilization in adsorption, hydrogen bonds, coordination bonds, and Van der Waal's forces are intertwined (Wang et al. 2009). However, some disadvantages are identified as a result of the binding's weakness. Desorption of the bound enzyme can occur as a result of changes in temperature, pH, ionic strength, or the presence of substrate, for example (Engasser and Horvath 1975). Furthermore, the sustenance does not selectively fix the enzyme; other irrelevant matter can also be adsorbed onto the environment, causing impediments such as denaturation of the enzyme to become inconsistent.

Chemical methods or irreversible

Entrapment

The enzymes can be surrounded with courses such as encapsulating an enzyme with a polyacrylamide gel (Klibanov 1983). The free diffusion of low molecular weight substrates is dealt with in this approach of immobilizing enzymes, and the final products and enzymes, which are considerably larger particles, cannot escape out of the beads (Mallardi et al. 2015). Several matrices are used to capture enzymes, including polyurethane foams, silastic gels, and starch gels (Jancsik, Beleznai, and Keleti 1982). There is no bond formation between the enzyme and the matrix, implying that the enzyme is exposed to a minimum of severe stresses. Though a wide range of enzymes can be immobilized using entrapment methods, only low molecular weight substrates can be employed in such performances. Because food systems include macromolecules, this could provide a check for their usage in food knowledge (Jun et al. 2019; Kilara et al. 1979). Academically, the pore size of the polymer should prevent enzyme leakage; nevertheless, in practice, a wide range of pore sizes are encountered, which can result in enzyme loss due to diffusion (Bayne, Ulijn, and Halling 2013; Califano and Costantini 2020).

Cross-linking

Bifunctional reagents, such as glutaraldehyde, were used to join intermolecularly the molecules of enzymes. Bifunctional compounds can contain two functional groups that are the same or different, as well as groups of reactivities that are different (Kim and Herr 2013; Kluger and Alagic 2004). The intermolecular associating enzyme molecules are inadequate, as the enzyme's support function, resulting in low events. Because enzymes are expensive biochemicals, inadequate use of enzymes in this technique results in a price increase for immobilized enzymes (Bedran-Russo et al. 2014).

Covalent bonding

The method of enzyme immobilization is based on the creation of strong covalent bonds between the functional groups on the carrier particle and the enzyme (Brena, González-Pombo, and Batista-Viera 2013). The amino groups (NH_2) of arginine or lysine, a carboxylic group (COOH) of glutamic acid or aspartic acid, a hydroxyl group (OH) of threonine or serine, and a sulfhydryl group (SH) of cysteine are all covered on the enzyme molecule's surface (Sulaiman et al. 2015).

Ionic bonding

The standard of a protein-ligand interface is ionic bonding, which is the basis of enzyme immobilization used in chromatography, such as -amylase on calcium phosphate (Ueland et al. 1993). This procedure is cost-effective since it is simple and affordable, and it results in minimal or no loss of enzyme or carrier molecule (Katchalski-Katzir and Kraemer 2000).

Entrapment of enzyme in support

The process of entrapment leads to enzyme inclusion within a polymeric structure. The enzyme is supported by allowing the substrate and yields to pass through (Rodriguez-Abetxuko et al. 2020). The support acts as a barrier to mass transfer and serious reaction kinetics (Benzinger, Becker, and Hüttinger 1996). There are numerous major approaches to entrapment, including ionotropic gelation of macromolecules with multivalent cations (alginate), temperature-induced gelation (agarose, gelatin), organic polymerization reaction by biochemical/photochemical (polyacrylamide), and precipitation from an immiscible solvent (polystyrene). On a polyacrylamide gel, -amylase or amyloglucosidase are two examples (Pillay et al. 1998; Bocchetta 2020).

Cross-linking of the enzyme with support

In this approach of enzyme immobilization, chemical bonding is used to connect an enzyme molecule to the support, resulting in the use of a three-dimensional structure. As the most common crosslinking agents, glutaraldehyde

Table 4. Potential applications of microbial lipases.

Industries	Applications of Products	Action
Food Industry		
Fats and oils	Cocoa butter, margarine, fatty acids, glycerol, mono- and diglycerides	Transesterification, hydrolysis
Meat and fish	Meat and fish product, fat removal	Flavor development
Health foods	Health foods	Transesterification
Food sauce	Mayonnaise, condiment, and whipping cream	Quality improvement
Beverages	Alcoholic beverages, e.g., sake, wine	Improved aroma
Bakery foods	Shelf life propagation	Flavor improvement
Dairy foods	Development of flavoring agents in milk, cheese, and butter	Hydrolysis of milk fat, cheese ripening, modification of butter fat
Agrochemicals	Herbicides such as phenoxypropionate	Esterification
Fuel industries	Biodiesel production	Transesterification
Pollution control	To remove hard stains and hydrolyze oil and grease	Hydrolysis and transesterification of oil and grease
Cosmetics	Act as emulsifiers and moisturizers	Synthesis
Pharmaceuticals	Production of various intermediates used in the manufacture of medicine	Hydrolysis of expolyester alcohols
Detergents industry	Removal of oils stains from fabrics	Hydrolysis of fats

bonds via its amino group, whereas diamines crosslink via carboxyl groups (Weinhold 2012; Zhou, Fan, and Chen 2016). The enzymes are well attached, therefore there are few chances of enzyme desorption using this method. Cross-linking, on the other hand, has the potential to create significant changes in the dynamic site of the enzyme molecule, which is a disadvantage of this method.

Using nanotechnology in immobilization

For enzyme immobilization, nanoparticles, nanofibers, nanotubes, nanopores, nanosheets, and nanocomposites are employed as nano scaffolds (Misson, Zhang, and Jin 2015). Using nanomaterials for enzyme immobilization has various advantages, including high enzyme loading, low mass transfer resistance, high mechanical strength, quick separation process, and better enzyme stability and activity thanks to the vast surface area. Nanocarriers were exposed to chemical and thermal accepting nanocatalysts with high enzyme loading and increased catalytic activity (Bilal et al. 2019). Because of their high surface-to-volume ratio, minimal mass transfer confrontation caused by the outer magnetic field, and ease of separation, magnetic nanoparticles are the best standard carriers for enzyme immobilization among all nanocarriers for enzyme loading (Johnson, Park, and Driscoll 2011; Husain et al. 2011).

Advantages of enzymes immobilization

Immobilization improves lipase stability at high temperatures, and immobilized lipase is more active than free lipase. The optimal reaction temperature for immobilized lipase shifts from 37-50 °C to 37-50 °C for free lipase (Carvalho et al. 2013). High temperatures have less of an impact on lipase activity, although they do improve thermal stability. Immobilized *Candida rugosa* lipase has an optimal temperature of 20-50 °C, while free *Candida rugosa* lipase has an optimum temperature of only 40-50 °C (Mokhtar et al. 2020: Lee et al. 2011). The electrostatic charges shifted after immobilization, causing the immobilized enzyme's optimum pH to shift to a mildly alkaline range (Kahveci and Xu 2011). The number of acidic groups increases after immobilization, making the enzyme more polyanionic appealing (Guzik, Hupert-Kocurek, and Wojcieszyńska 2014). In alkaline settings, immobilized lipase has higher activity than free lipase. Immobilization improves enzyme characteristics by enhancing enzyme stiffness and heat resistance (Matsumoto and Ohashi 2003). It also causes the enzyme to have limited structural flexibility, which is evidenced by an increase in the optimal temperature and stability against inactivation. Several industrialized methods necessitate high temperatures and better thermal performance of immobilized enzymes, which serve as the foundation for enzyme applications (Sirisha, Jain, and Jain 2016).

Potential applications of microbial lipases

In the processing of food, fats, and oils, the synthesis of fine chemicals and pharmaceuticals, detergents and degreasing formulations, paper manufacture, pharmaceuticals, biosensors, pulp and paper, textile production of cosmetics, industry, etc. Lipases are widely used (Sharma, Chisti, and Banerjee 2001). Major potential applications of microbial lipases are summarized in Table 4 (Aravindan et al. 2006).

Lipases in food industry

Enzymes are found in a wide range of products used by humans and animals regularly. It can be utilized as a dietary additive by changing the location of fatty acid chains in the glyceride and substituting one or more fatty acids with new ones (Alves-Bezerra and Cohen 2017; Henry 2009). Lipase can alter the properties of lipids, boosting the flavor of some foods, improving their texture, prolonging the shelf life of others, and increasing the softness of others, as well as improving food quality (Stanhope 2012; Turan and Erkoyuncu 2012).

In flavouring and aroma

Microbial enzymes generate a wide range of aromatic and flavoring compounds used in the food industry. Salad dressings, dairy, confectionery, and bread products all use lipases

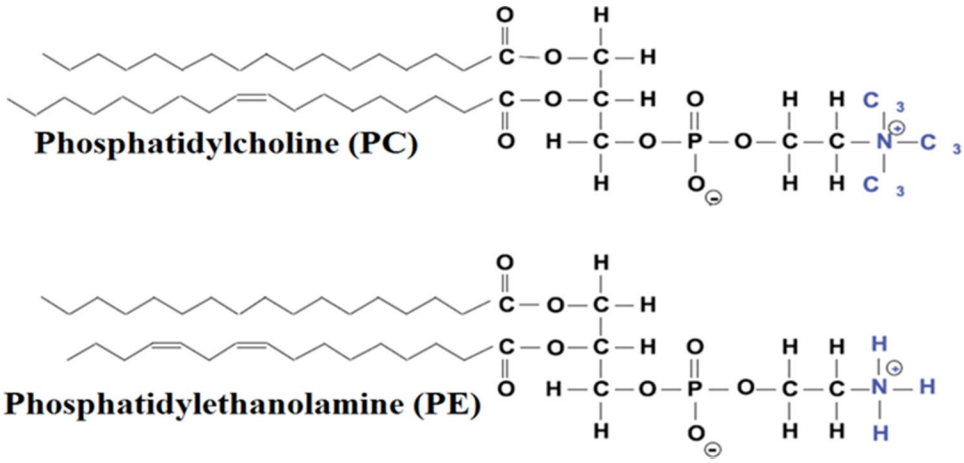

Figure 3. Phospholipids chemical structure in egg yolk (Chandra et al. 2020b).

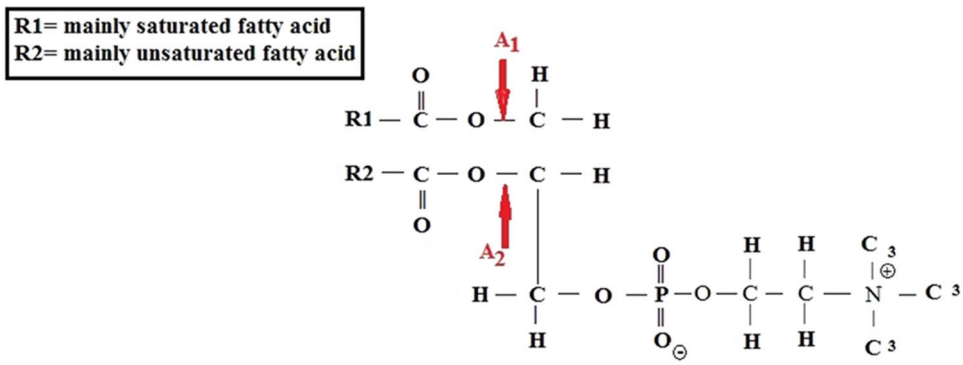

Figure 4. Phospholipases action in A1 and A2 positions (Guerrand 2017).

to flavor and aromatize them. Lipases used in the tea industry for the production of the flavor, the concentration of polyunsaturated fatty acids increases while the lipid composition decreases. It's also used to provide a distinct fragrance in alcohol manufacturing. Short-chain fatty acids were converted into esters by lipase enzymes, which gave food its flavor. Processing variables like pH, temperature, amount, and emulsion content all influence the level of flavor and scent (Rajendran, Palanisamy, and Thangavelu 2009). Lipase can be used to change the flavor of a cake, cookie, dough, cheese, or confectionery, among other things.

In human milk fat substitutes

Several lipids are contained in human milk fat (HMF), including palmitic (20-30%), stearic acids (5.7-8%), linoleic (7-14%), and oleic (30-35%) (Sánchez-Hernández et al. 2019). Unlike vegetable oils and cow's milk fat, palmitic acid is the predominant saturated fatty acid in HMF and is esterified at the sn-2 position of the TAGs (Zou et al. 2013; Sahin, Akoh, and Karaali 2005). Unsaturated fatty acids are found at the exterior positions (Bracco 1994). The profile of HMF fatty acid has a significant impact on its digestion and intestinal absorption in babies (Ghosh et al. 2016). Human milk fat alternatives (HMFS) have been produced using sn-1, 3 lipase-catalyzed acidolysis of tripalmitin, butterfat, palm oil, palm stearin, or lard (rich in palmitic acid in the sn-2 position) and free fatty acids (FFA) from various sources (Soumanou, Pérignon, and Villeneuve 2013). The commercial Betapol® product is industrialized utilizing IOI Loders Croklaan, by acidolysis between lard and soybean fatty acids, catalyzed by sn-1, 3 specific lipases from *Rhizomucor miehei* (Lipozyme® RM) (Haraldsson et al. 1989; Xu 2000).

Lipases in egg processing

In the food sector, eggs provide functional ingredients with a variety of qualities, including foaming, gelation, emulsification in batters and mayonnaise, and enhanced texture of baked items (Foegeding, Luck, and Davis 2006). The emulsifying capabilities of eggs are also due to their lipids (Anton 2013). Emulsification of egg yolks in dressings: The global production of emulsified dressings is projected to be 3 million metric tons per year, with around 15, 0000 metric tons of egg yolks consumed in the process (Alu'datt et al. 2017; Miranda et al. 2015). One-third of the market for emulsified dressings is centered in Russia and Eastern European nations. Nestlé, Kraft, and Unilever operate in a highly industrialized market with worldwide players (Ene-Obong and Carnovale 1992). The egg yolk was created as a complicated oil-water emulsion with 50 percent water, 32 percent

lipids, and 16 percent protein (Mine 1998). Egg yolk contains phosphatidylethanolamine; roughly 1/3 of the lipids are phospholipids, with phosphatidyl-choline (PC) (Figure 3), accounting for about 80% of the total. The emulsion stability was improved due to the enzymatic conversion of egg yolk phospholipids into lysophospholipids (van Hoogevest and Wendel 2014; Pichot, Watson, and Norton 2013).

Performing at several positions enzyme constructers have industrialized dissimilar phospholipases (PL) (Figure 4). Microorganisms (Maxapal A2, DSM, NL) utilized for egg processing, and lipases derived from hog pancreas (Lipomod 699, Biocatalysts, UK) (Bjurlin, Bloomer, and Haas 2001). Using pork pancreas lipases, nearly 80% of phospholipids are converted to lysophospholipids after 1 hour. Although the enzyme is of animal origin (vegan consumers, Halal/Kosher requirements), its use is restricted (Fuglsang et al. 1995). The high conversion yield is also influenced by cloned and articulated *Aspergillus niger* lipases (such as DSM Maxapal A2), although numerous industrial players are opposed to depleting enzymes produced by reformed microbes.

Lipases in bakery and confectioneries

In most bakers' goods, enzymes are the most important element. Due to the controlled use of chemical additions in the production of bread and other fermented foodstuffs (Numan and Bhosle 2006). Enzymes are predicted to play an increasingly bigger role in baking shortly. Wheat flour is the most important ingredient in the production of baked products such as bread, cake, pastries, biscuits, crackers, cookies, pies, and tortillas, as well as a vital source of enzyme substrates (Villarino et al. 2016). The first generation is 1, 3-specific, preferring removing fatty acids from positions 1 and 3 in TAG (Beisson et al. 2000; Grönke et al. 2005). Lipases (TAG lipases) improve dough rheology, increase dough strength and stability, and hence improve dough machinability. Lipases function simultaneously on TAG, diacylgalactolipids, and phospholipids to generate more polar lipids, higher dough stability, and a fine, homogeneous bread crumb structure (Huang et al. 2019). Lipase enhances the volume and crumb structure of high fiber white bread by increasing the expansion of the gluten network, wall thickness, and cell density.

Lipases in cocoa processing

Cocoa butter has a melting point of 37 °C and contains palmitic and stearic acids. It melts in your mouth, giving you a cooling sensation (Bloomer, Adlercreutz, and Mattiasson 1990). Unilever patented this invention in 1976 to create a cocoa butter substitute utilizing immobilized lipase (Owusu-Ansah 1994; Ewens, Metilli, and Simone 2021). *Rhizomucor miehei* commercializes the immobilized lipases which form the transesterification reaction. In this technology, palmitic acid will be replaced by stearic acid, resulting in stearic-oleic-stearic triglyceride.

Processing of edible fats and oils

Immobilized lipase is widely utilized in the oil and fat business, and is primarily employed in the processing of fats and oils to enhance flavor (Murty, Bhat, and Muniswaran 2002). The immobilization technique improves the enzyme's stability and activity; immobilized lipase is preferred over free enzyme (Facin et al. 2019; Liu et al. 2012). The immobilized version of the enzyme can be reprocessed. The transesterification reaction induced by *R. miehei* is carried out with immobilized lipase. Palmitic acid is substituted with stearic acid in this method (Sultan and Sokolove 2001). Immobilized lipase is employed for the esterification of functionalized phenols and the production of antioxidants, and it is also castoff in sunflower oil due to its lipophilic nature (Zoumpanioti et al. 2010). Trans-fat-free oils are also made with immobilized lipases. Although soy oil has low oxidative stability, it does contain a significant amount of unsaturated fatty acids (Song et al. 2015). As a result of partially hydrolyzed activity, trans-fat was formed, which harmed health (Klonoff 2007; Tyburczy, Mossoba, and Rader 2013). Enzymatic interesterification is possible at very low aqueous medium concentrations. The immobilized enzyme is also utilized to create zero or low trans-fat oil, shortening, and cooking oil using corn, sunflowers, and/or soy oil as a foundation (Farfán et al. 2015; Colón-Ramos, Monge-Rojas, and Campos 2014).

Lipases in the cheese industry

Following the settling whey was a fractional or full mixture of coagulating milk, cream, skimmed milk, or partially skimmed milk, buttermilk, or its fresh and mature products known as cheese. The enzymatic process is the most prevalent method of milk coagulation; there are two types of cheese manufacturing processes: acidification and enzymes that coagulate milk (Dalgleish 1993; McMahon, Brown, and Ernstrom 1984). The creation and ripening of this sort of cheese are done by lipase-like enzymes, and the cured and matured cheese is not ready to eat right away. The texture and flavor of fat have an important impact on the cheese properties of numerous milk ingredients. Based on the composition of the base of fat, cheeses can be classified as having (less than 25 percent) less fat, (25-45 percent) moderate fat, (45-60 percent) loaded fat, and (> 60 percent) more fat (Moatsou et al. 2019).

Cheese flavor improves throughout maturation via primary (lipolysis, proteolysis, metabolism of residual lactose, lactate, and citrate) and secondary (metabolism of fatty acids and amino acids) biochemical pathways due to the production of a wide spectrum of complexes (Forde and Fitzgerald 2000; McSweeney 2004). Mold-ripened cheeses such as Camembert, Brie, Roquefort, and Stilton are distinguished by a wide range of lipolysis. In such cheeses, both proteolysis and lipolysis leave a distinct flavor and scent (Dugat-Bony et al. 2015; Brennan et al. 2004). Lipases derived from a variety of sources catalyzed cheese by lipolysis, primarily by enzymes found in rennet adhesive, in the creation of cheese varieties such as Provolone, old-fashioned Greek Feta, and

Italian Pecorino (Rani and Jagtap 2019; Schirone et al. 2012). Secondary reactions result from the degradation of fatty acids and amino acids in the production of volatile taste molecules (Ianni et al. 2020; Wang et al. 2020). Lipolysis has an important part in the production of large quantities of short-chain volatile fatty acids in EMC taste, which is linked to aroma intensity and flavor (Kilcawley et al. 1998; Mohebbi et al. 2008; Lee et al. 2007).

The EMC emulsification course, which includes substrate-assisted lipolysis, provides an ideal environment for lipases by increasing the oil-water contact (Li and McClements 2011). Which has a long-term preference for triglycerides, particularly those found near an oil-water interface, over free solutions, which can work up to 1000 times quicker (Armand et al. 1999; Golding et al. 2011). The significance of uncontrolled lipolysis is the increase of rancidity or soapiness due to an imbalance between short- and long-chain fatty acids (Sobczak, Blindauer, and Stewart 2019; Crielly, Logan, and Anderton 1994). Using the lipolytic process with esterase and lipase activities to prevent the off-flavor (Kilcawley, Wilkinson, and Fox 2001). A variety of EMC flavors are used to make blue, Swiss, Gouda, Mozzarella, and Parmesan cheeses (Fox 1993). Cheddar EMC is particularly important because it can be found in a variety of supposed Cheddar flavors and intensities from various manufacturers. Bacterial enzymes play a key role in the formation of taste in surface-ripened cheeses; these smear bacteria hydrolytic activities are involved in the production of cheese flavor components (Williams, Beattie, and Banks 2004) and this is determined by on profile of enzyme of pressures elaborate in the starter culture (Stefanovic et al. 2018; Bockelmann 2000; Rubio et al. 2014).

Lipases in the fish and meat industry

Lipase is also utilized in the meat and fish industries to generate lean meat. It is also used to improve its suitability for the fermentation of meat products to improve the superiority of fermented sausages. The aggregation of unsaturated fatty acids (n-3 PUFA) and the hydrolysis of fish oil lipases are employed (Yan et al. 2011).

Applications of lipases in the oleo-chemical industry

In addition to their natural role of hydrolyzing carboxylic ester bonds, lipases can catalyze esterification, inter-esterification, and transesterification processes in non-aqueous conditions (Yadav and Devendran 2012; Nishio et al. 1989). In the modern oleo chemical sector, the use of immobilized lipases to initiate diverse reactions such as hydrolysis, alcoholysis, and glycerolysis, as well as the usage of mixed substrates, has expanded significantly (Arana-Peña et al. 2020). As a result of the application of immobilized enzymes, the practices of continuous running and high production are confirmed. As a result, fat splitting and alteration can be accomplished without a major investment in expensive equipment or the expenditure of enormous amounts of thermal energy (Chapman, Ismail, and Dinu 2018; Ansari and Husain 2012). Lipases have a wide range of applications because they save energy and reduce heat deterioration during hydrolysis, glycerolysis, and alcoholysis (Arbige and Pitcher 1989). Ethyl caprylate, a sweet fruity-flowery flavoring produced from *Bacillus subtilis* EH37 strain employing immobilized lipase, is a very valuable tool for food and beverage flavorings. Using lipase, *Streptomyces thermocarboxydus* ME168 strain was found to create sugar esters from a variety of substrates including glucose, fructose, and different vinyl esters (Siebenhaller et al. 2018).

Lipase in bioremediation

Lipids and carbohydrates are the principal waste materials produced by the food industry, and the formation of oily coatings on aquatic surfaces poses a threat to the ecosystem. Because of the emulsification with organic materials, the oily coating clogs and prevents oxygen diffusion (Baloch and Hameed 2005). The cytotoxic potential of cooking oil waste can be reduced by enzymatic activity based on lipase, which has well-known environmental consequences (Okino-Delgado et al. 2017). Lipases catalyze a wide range of lipid changes because of their broad substrate affinity and great temperature and solvent stability (Kanmani, Aravind, et al. 2015). The effect of immobilized lipases such as *P. aeruginosa, Staphylococcus epidermidis*, and *Bacillus subtilis* on the removal of stored lipids and grease from wastewater pipelines has been evaluated and is known (Hatzisymeon, Kamenopoulos, and Tsoutsos 2019).

Filamentous fungus-like *Penicillium chrysogenum, Penicillium cyclopium, Penicillium simplicissimum, Penicillium expansum, Candida rugosa, Aspergillus fumigatus, Trichoderma* sp. (Bancerz et al. 2005; Nowak, Kucharska, and Kamiński 2019; Mehta, Bodh, and Gupta 2018), and bacterial strains like *Bacillus subtilis*, *Pseudomonas* sp. (Singh et al. 2018; Hu et al. 2018). The toxicity of jatropha seed cake (JSC) is produced in large quantities as a by-product in the biodiesel industry. The AAU2 lipase enzyme from *Pseudomonas aeruginosa* degraded and detoxified phorbol ester (phorbol 12-myristate 13-acetate) using JSC as a substrate (Muhamad et al. 2019).

The disposal of grease waste poses a severe challenge and poses a hazard to the ecosystem because it is non-biodegradable. Waste grease has been thrown in littered sites or sewages in numerous countries without any preparation (Jayathilakan et al. 2012; Toldrá, Mora, and Reig 2016; Kanmani, Aravind, et al. 2015). Grease wastes induce a decrease in cell-aqueous phase transfer rates, reduced sedimentation, the creation of floating sludge, clogging, and the appearance of foul aromas in effluents (Chandra and Singh 2014). The lipase-producing activity of *P. chrysogenum* SNP5 is employed for grease cleanup (Yeo, Nihira, and Yamada 1998).

P. putida GPo1 alkB; *Acinetobacter* spp. alkM; *Rhodococcus* spp. alkB1, *Rhodococcus* spp. alkB2, *P. putida* xylE, *P. putida* ndoB, and *Mycobacterium* sp. strain PYR-1 nidA are indigenous cold-adapted microbes that degrade n-alkane (Hasan, Shah, and Hameed 2006). *Pseudomonas protegens* BC2-12,

Pseudomonas protegens CHA0, *Pseudomonas protegens* Pf-5, *P. fluorescens* A506, and Pf0-1, *Pseudomonas chlororaphis* lipase producing bacteria are used in the biodegradation of polyurethanes (PUR) (Wilkes and Aristilde 2017; Chandra, Enespa, and Singh 2020a). *P. aeruginosa* has been found to produce large levels of extracellular lipase, which aids in the biodegradation of polyesteramides and aromatic-aliphatic polyesters. One of the first enzymes on PUR was PueB lipase, which was discovered in *Pseudomonas chlororaphis* (Wilhelm, Tommassen, and Jaeger 1999; Pathak and Navneet 2017). The industrial bacterial residue of *Paracoccus* sp. strain KDSPL-02 was used to biodegrade penicillin G in an extended bed adsorption bioreactor (EBAB) (Nerurkar et al. 2013).

Lipase in detergent engineering

Pseudomonas aeruginosa AAU2 lipase enzyme showed significant stability in the presence of commercial detergents such as Ezee® and Wheel®. Ohgiya et al. 1999, used a detergent made from *Bacillus sonorensis* and lipase to replace corn-oil stains on un-dyed cotton fabric. Cold-active lipases can be used as additions in detergent formulations and in an organic mixture with a chiral intermediate to wash garments at low temperatures in the laundry (Unni et al. 2016). An alkaline and thermotolerant lipase produced by *Pseudomonas aeruginosa* strain BUP2 (Olusesan et al. 2009) are employed in the detergent business with efficiency and high specific activity. Lipo Prime ® is a lipase-containing detergent also made by them (Hemlata, Uzma, and Tukaram 2016). *Trichosporon asahii* MSR 54 produced an alkaline lipase, which was used to develop a presoak formulation for the removal of oil stains at room temperature (Guncheva and Zhiryakova 2011).

Lipases in the medical sector

Bacillus lipases can be employed in the pharmaceutical industry for the manufacture of enantiopure substances because of their characteristics. *Staphylococcus* lipase produces antioxidants such as tyrosol acetate, propyl gallate, and eugenol benzoate (Matull, Pereira, and O'Donohue 2006). When tuberculosis (TB) is detected, lipase can be utilized as a diagnostic tool. The level of lipase in blood serum can be utilized to detect acute pancreatitis and associated wound (McCutcheon 2000). A bile duct obstruction or an overdose of alcohol can cause pancreatitis (Hazem 2009). Lipases are employed in the creation of hair waving as an ingredient of topical anti-obese creams and for the treatment of malignant tumors since they act as TNF activators (Aghdassi et al. 2008). The lovastatin medication, which is made by *Candida rugosa* lipase, lowers serum cholesterol levels. Diltiazem hydrochloride is made via asymmetric hydrolysis of a crucial intermediate 3-phenylglycidic acid ester from *S. marcescens* lipase and is commonly used for coronary vasodilation (Gácser et al. 2007).

Lipases in pathogenicity mechanisms

Lipase 8 (LIP8) genes play an important role in *Candida albicans* pathogenicity. The mutant's deficit in the LIP8 gene was less virulent expressively in lipid-containing media lowering LIP8 expression observed in lowered growth and a murine intravenous contagion way (Toth et al. 2017; Fourie et al. 2018). Lip5 and Lip8 from *Candida albicans* promote host-pathogen contact (Chin et al. 2016). *Candida* species produce hemolysins, which decrease hemoglobin and extract elemental iron from host cells (Mayer, Wilson, and Hube 2013). Hemolysins were chosen as essential virulence factors because they may aid the pathogen's survival and persistence in the host. *Candida albicans*, regarded as a versatile pathogen implicated in oral, vaginal, and systemic infections, is responsible for a greater percentage of death in the United States due to infection in the circulation (Naglik, Richardson, and Moyes 2014; Chaffin 2008). Lip5, Lip6, Lip8, and Lip9 lipases were released by *C. albicans*, and they can allow the organism to attach and colonize in people after infection (Stehr et al. 2004; Park et al. 2019; Lemieux and Simard 1991).

Lipases mediated transesterification ergosterol manufacturing

Bacillus subtilis, Pseudomonas fluorescens, and *Aeromonas hydrophilia* are dairy food putrefaction bacteria that have produced unfavorable changes in the quality of food and cosmetics, as well as affecting the market (Machado et al. 2017). Psychrophilic *P. fluorescens* and *A. hydrophilia*, as well as thermophilic *Bacillus subtilis*, change the texture, off-flavor, and cause bitter odor in a variety of dairy products (Torbeck et al. 2011). *Burkholderia cepacia*, as a potential pathogen, caused many skin disorders as a result of an infection in creamy cosmetic goods (Akinboyo et al. 2018; Tavares et al. 2020). Several nosocomial outbreaks for immunocompromised individuals with cystic fibrosis and chronic granulomatous illness in health care facilities have been linked to *Burkholderia cepacia* complex (Bcc) infection (McMenamin et al. 2000). Ergosterol is used as a biomaterial substance in food and cosmetics manufacture because it has no antagonistic clinical effects with cholesterol and has various useful functional properties (Corrêa et al. 2017). However, because ergosterol is simply crystalline and has low oil solubility, it is difficult to employ in the business (Kuksis and Beveridge 1960). This difficulty can be remedied by combining fatty acids with ergosterol. Ergosterol oleate (EO), ergosterol linoleate (EL), and ergosterol linolenate (ELn) were produced with the help of *Proteus vulgaris* K80 lipase (He, Zhu, and Chen 2018; Park and Kim 2020). The microorganisms that cause putrefaction in dairy and cosmetics can be inhibited by these unsaturated fatty acid ergosterol esters (FAEEs) (Badellino et al. 2005).

Lipases use in a diagnostic tool

Lipases are also important therapeutic targets or enzyme markers in the medical field. Their presence or increasing

levels can be utilized to identify specific contagions or diseases and can also be employed as diagnostic tools. In the enzymatic resolution of blood triglycerides, glycerol lipases are used, with an enzyme-linked colorimetric response that is sequentially resolute (Treacy et al. 2001). The level of lipases in the blood serum can be used as an analytical method to detect situations such as pancreatic damage and acute pancreatitis illnesses (Kivanc, Yilmaz, and Demir 2011). A large number of virulence-related genes *Aeromonas* bacteria are found in drinking water, so it's important to look at as many different samples as possible to better understand the health risks these bacteria may pose (Sen and Rodgers 2004). The virulence characteristics of *Aeromonas* bacteria indicate that drinking water is a source of potentially harmful *Aeromonas* bacteria cleaned from municipal water. Rasmussen-Ivey et al. (2016) found that *P. acnes, Corynebacterium acnes*, and *Staphylococcus aureus* are pathogenic bacteria and that lipase from these bacteria influences skin rash in acne patients (Moradali, Ghods, and Rehm 2017). Many enzymes, including lipases, are discovered as biological materials subsidizing the pathogenicity between the virulence factors by an opportunistic bacterium known as *P. aeruginosa* to produce and secrete numerous different virulence factors (Rasamiravaka et al. 2015; Vasil 1986).

Lipases in pharmaceuticals

Aryl aliphatic glycolipids, citronellol laurate from citronellol and lauric acid, and ethyl esterification of docosahexaenoic acid to ethyl docosahexaenoic acid to ethyl docosahexaenoic acid to ethyl docosahexaenoic acid are the cold-active lipases utilized in industrial (Ota, Sawamoto, and Hasuo 2000; Kauffmann and Schmidt-Dannert 2001; Ugur and Boran 2014). The fatty acid chain length of an ester was selected by *Bacillus* lipases, although only a small number of enzymes showed positional specificity (Ranjan et al. 2016; Horchani et al. 2010). *Bacillus* lipases can be discarded in the pharmaceutical industry for the synthesis of enantiopure complexes because of these features (Contesini et al. 2020). *Staphylococcus* lipase is used to produce antioxidant qualities such as tyrosol acetate, propyl gallate, and eugenol benzoate (Lim and Lee 2012). In the case of tuberculosis (TB) detection, lipase can be employed as a diagnostic tool. *Mycobacterium tuberculosis* lipase is used to assess for infection with good specificity and sensitivity (Nyendak, Lewinsohn, and Lewinsohn 2009; Li and Lo 2015). They have immobilized because of their regio-, chemo-, and enantioselectivity. CalB is a catalyst and pharmacological active ingredient in the synthesis of Odanacatib (Bovara et a., 1993). Chronic liver disease is caused by the hepatitis C virus (HCV). Sofosbuvir is used to treat hepatitis C, an infectious condition (Richmond, Dunning, and Desmond 2007; Krystallis et al. 2012). Cirrhosis of the liver manifested itself as liver failure, gastric varices, and, ultimately, malignancy. Racemic atenolol from *Candida antarctica* is kinetically resolved using lipase (Siebenhaller et al. 2017). The atenolol of enantiomeric resolution was used as an acrylate agent and an organic solvent in a transesterification procedure that used vinyl acetate as the reaction medium (Faraldos et al. 2011; Carvalho et al. 2006). The lipase from an *Aspergillus niger* strain produced favorable results in the enantiomeric resolution of racemic ibuprofen (da Silva et al. 2009; Lee and Shuler 2007).

When the enzyme catalyzes enantiomerical esterification of R-ibuprofen, the enantiomer results in a higher ratio of S-ibuprofen (Ong, Kamaruddin, and Bhatia 2005; Zhangde, Jianhe, and Jiang 2007). In addition, *Serratia marcescens* lipases produce chiral drug precursors that are hydrolyzed enantioselectivity (Zhao et al. 2008; Zanger and Schwab 2013). Enzymatic acylation of amines has been abandoned for the training of pharmacologically interesting -substituted isopropyl amines (Nordwald, Armstrong, and Kaar 2014; Andriianova, Biglova, and Mustafin 2020). The inclusion of methyl groups on the nitrogen atom or methoxy groups on the aromatic ring is known to increase the drug's effects (Gibbons 2012; Abdelraheem et al. 2019). All of these types solidify in the sphere of organic separation and pharmaceutical commerce with the employment of lipases striking for biotechnological uses on the population's strength with the prospect of a progressive effect (Salihu and Zahangir Alam 2015; Reetz 2002). To manufacture enantiomerically pure secondary alcohol, there is multiple instances structure that encloses numerous chiral pharmaceuticals or intermediates that may be accepted out for the separation of pharma medications by acylation of alcohol or hydrolysis of esters utilizing lipases in organic solvents (Kato et al. 2007; Liu et al. 2010; Weissfloch and Kazlauskas 1995). As a result, *Pseudomonas cepacia* lipase (PSL) used enzymatic acylation of halogen alcohols or cianoalcohols to disclose a dynamic kinetic resolution of azidoalcohols for the creation of (S) -propranolol (Fernandez and Khiar 2003). This mixture of substances has more interest as calcium antagonists and is usually employed in the pharmaceutical business as a target in the creation of optically pure 1, 4-dihydropyridine derivatives, with enantiomerically pure 2-methoxy-2-phenyl ethanol, observed as a desirable chiral auxiliary. *Candida antarctica* lipase B (CAL-B) can be used to resolve this primary alcohol after an E of 47 using a variety of lipases through enzymatic acylation. Paroxetine hydrochloride is a selective serotonin (5-HT) reuptake inhibitor that is used as an antidepressant (Davis et al. 2016).

Lipases used in biosensors

The breakdown of fats, oils, and phospholipids is catalyzed by the lipase and phospholipase enzymes, respectively. They can be used to detect a few diseases or contagions, such as high triglycerides or pancreatitis, and their presence or rising levels can be employed as diagnostic tools (Edwards and Tchounwou 2005). Pesticide residues containing organophosphorus (OP), commonly known as p-nitrophenyl pesticides (methyl parathion), are neurotoxic and difficult to remove from contaminated water (Ma et al. 2018; Cheng et al. 2020; Zehani et al. 2015). For the detection of methyl parathion direct and rapid lipase@ZIF-8/chitosan/glassy

carbon electrode (GCE) and lipase @amine-modified ZIF-8(An-ZIF-8),/chitosan/GCE biosensors have been developed (Senbua, Mearnchu, and Wichitwechkarn 2020; de Moura Barboza et al. 2019; Ribeiro et al. 2011). Lipase@ ZIF-8/chitosan/GCE and lipase@An-ZIF-8/chitosan/GCE are methyl parathion modified biosensors with a wide linear range, good reproducibility, recovery, and a detection limit as low as 0.28 M (Coelho, Silva, and Orlandelli 2021; Yang et al. 2003). The carbendazim (CBZ) insecticide was determined using lamellar zinc hydroxy nitrate adorned gold nanoparticles immobilized with *Ceratobasidium* sp.2 lipases and then mixed into carbon paste using multi-walled carbon nanotubes (CNTs) (Chen et al. 2019). Potentiometric biosensors based on *Candida rugosa* lipase have been developed for the analysis of organophosphorus pesticides such as methyl-parathion and tributyrin (Kartal, Ali Kilin, and Timur 2007).

Urea in milk can also be detected with potentiometric biosensors (Trivedi et al. 2009). The urease enzyme was immobilized utilizing a polymer matrix of polyethyleneimine and poly (carbamoyl) sulfonate to capture it on an ion-sensitive membrane. The detection limit and average slope of the biosensor in the linear range were 2.510 5 mol/L and 51.7 0.5 mV/decade, respectively. Allergens in foods like milk and dairy products have sparked worries about their safety. Eissa et al. (2012) developed a label-free electrochemical immunological biosensor with a detection limit of 0.85 pg/mL for the detection and quantification of -lactoglobulin, a milk allergy. A chemical process and an organic film (graphene-modified screen-printed electrode) were used to make the biosensor.

Another analyte to be concerned about in milk and dairy products is bisphenol A (BPA). BPA is commonly utilized as a monomer in the manufacture of polycarbonate plastics and epoxy resin, which are commonly used to construct a variety of food packaging materials such as water and dairy bottles, as well as coating material for processed food cans (Kang and Kondo 2003; Lim et al. 2009; Goodson et al. 2004). Alkasir, Ornatska, and Andreescu (2012) developed a BPA-detecting enzyme-based nanoparticle functionalized electrochemical biosensor.

Melamine is a high-nitrogen organic chemical that has been illegally added to milk and its products, causing significant poisoning in humans (kidney damage). A melamine biosensor based on ionic liquid/nanoparticles/chitosan and a modified gold electrode was developed for the determination of melamine in milk and its products (Rovina, Siddiquee, and Wong, 2015). Melamine may be detected using a melamine biosensor with an exposure limit of 9.6×10^{-16} M and a concentration range of 9.6×10^{-15} – 3.3×10^{-3} M. As a result, during food safety inspections, it can be used to look for melamine in milk products (Alizadeh et al. 2022).

Lipases in the pulp and paper industry

The paper and pulp business processes a large amount of lignocellulosic biomass per year. Triglycerides and waxes, sometimes known as hydrophilic components of wood (Pitch), combine to produce some challenges in the pulp and paper manufacturing process (Gutiérrez, Río, and Martínez 2009). One of Japan's largest paper mills, Nippon, developed this technology to regulate impurity from the wood pitch by affecting their hydrolysis (90 percent) utilizing C. rugosa fungal lipase (Patel et al. 2019). To avoid the detrimental effects of the negative characteristics on the operation of the papermaking equipment Anion residue and bitumen deposition can also be treated with lipase (Demuner et al. 2011). To increase manufacturing capacity and quality (Horchani et al. 2012), it may be necessary to eliminate the esters from pulp lipase.

Conclusions and future perspective

Lipases should be able to withstand multiple reaction cycles in biotechnological applications. This stability can be improved through immobilization methods and catalytic characteristics, resulting in increased catalyst efficiency. Protein adulteration is reduced or eliminated when enzyme purification is used. Immobilized lipase enzymes help a lot in the industrialized techniques where they're used currently. Oversimplification of the process reduced environmental impact, and a unique ecological approach to biochemical separation are all well-known benefits of aid. In well-established production techniques such as cocoa butter analogues goods, amino acids, bakery and confectionary, medications, and pharmaceuticals, the food sector uses the most immobilized enzymes. Enzyme catalysis on a commercial scale has been used in a variety of industries, including medicines and food, with recent trends in biofuel production and natural gas conversion. Immobilized enzymes will be employed more widely in the future, bringing in the era of enzyme technology.

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Abbreviations:

CBZ	Carbendazim
EMC	Enzyme-modified cheese
EMDI	Enzyme-modified dairy ingredients

EL	Ergosterol linoleate
ELn	Ergosterol linolenate
EO	Ergosterol oleate
FFA	Free Fatty Acids
GCE	Glassy carbon electrode
HCV	Hepatitis C virus
HMF	Human Milk Fat
OP	Organophosphorus
PC	Phosphatidyl-Choline
PUR	Polyurethane
TB	Tuberculosis

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References

Abdala, A. F., A. P. Gallardo, L. G. Olvera, and E. M. E. Silva. 2017. Hydrolysis of carotenoid esters from *Tagetes erecta* by the action of lipases from *Yarrowia lipolytica*. *Bioresources and Bioprocessing* 4 (1):5–12. doi: 10.1186/s40643-016-0131-7.

Abdelraheem, E. M., H. Busch, U. Hanefeld, and F. Tonin. 2019. Biocatalysis explained: From pharmaceutical to bulk chemical production. *Reaction Chemistry & Engineering* 4 (11):1878–94. doi: 10.1039/C9RE00301K.

Abramić, M., I. Leščić, T. Korica, L. Vitale, W. Saenger, and J. Pigac. 1999. Purification and properties of extracellular lipase from *Streptomyces rimosus*. *Enzyme and Microbial Technology* 25 (6):522–9. doi.org/ (99)00077-0 doi: 10.1016/S0141-0229.

Adachi, D., S. Hama, K. Nakashima, T. Bogaki, C. Ogino, and A. Kondo. 2013. Production of biodiesel from plant oil hydrolysates using an *Aspergillus oryzae* whole-cell biocatalyst highly expressing *Candida antarctica* lipase B. *Bioresource Technology* 135:410–6. doi: 10.1016/j.biortech.2012.06.092.

Adamczak, M., and W. Bednarski. 2004. Enhanced activity of intracellular lipases from *Rhizomucor miehei* and *Yarrowia lipolytica* by immobilization on biomass support particles. *Process Biochemistry* 39 (11):1347–61. doi: 10.1016/S0032-9592(03)00266-8.

Adlercreutz, P. 2013. Immobilisation and application of lipases in organic media. *Chemical Society Reviews* 42 (15):6406–36. doi: 10.1039/C3CS35446F.

Adrio, J. L., and A. L. Demain. 2014. Microbial enzymes: Tools for biotechnological processes. *Biomolecules* 4 (1):117–39. doi: 10.3390/biom4010117.

Aghdassi, A., J. Mayerle, M. Kraft, A. W. Sielenkämper, C.-D. Heidecke, and M. M. Lerch. 2008. Diagnosis and treatment of pancreatic pseudocysts in chronic pancreatitis. *Pancreas* 36 (2):105–12. doi: 10.1097/MPA.0b013e31815a8887.

Ahmad, M., H. Melanie, P. Harald, and S. Helmut. 2014. Protein expression in *Pichia pastoris*: Recent achievements and perspectives for heterologous protein production. *Applied Microbiology and Biotechnology* 98 (12):5301–17. doi: 10.1007/s00253-014-5732-5.

Ahn, J. H., J. G. Pan, and J. S. Rhee. 1999. Identification of the tliDEF ABC transporter specific for lipase in *Pseudomonas fluorescens* SIK W1. *Journal of Bacteriology* 181 (6):1847–52. doi: 10.1128/JB.181.6.1847-1852.1999.

Akimoto, M., M. Izawa, K. Hoshino, K.-I. Abe, and H. Takahashi. 2003. Lipase-catalyzed interesterification of soybean oil with an ω-3 polyunsaturated fatty acid concentrate prepared from sardine oil. *Applied Biochemistry and Biotechnology* 104 (2):105–18. doi: 10.1385/ABAB:104:2:105.

Akinboyo, C. I., A. C. Sick-Samuels, E. Singeltary, J. Fackler, J. Ascenzi, K. C. Carroll, Y. Maldonado, R. B. Brooks, I. Benowitz, L. E. Wilson, et al. 2018. Multistate outbreak of an emerging *Burkholderia cepacia* complex strain associated with contaminated oral liquid docusate sodium. *Infection Control and Hospital Epidemiology* 39 (2):237–9. doi: 10.1017/ice.2017.265.

Albayati, S. H., M. Malihe, I. S. N. Hasmah, M. A. M. Shukuri bin, T. A. Leow, M. S. F. Binti, M. N N. D. Binti, and R. A. R. R. N. Zaliha. 2020. Main structural targets for engineering lipase substrate specificity. *Catalysts* 10 (7):747. doi: 10.3390/catal10070747.

Alcazar-Valle, M., A. Gschaedler, H. Gutierrez-Pulido, A. Arana-Sanchez, and M. Arellano-Plaza. 2019. Fermentative capabilities of native yeast strains grown on juices from different Agave species used for tequila and mezcal production. *Brazilian Journal of Microbiology: [Publication of the Brazilian Society for Microbiology]* 50 (2):379–88. doi: 10.1007/s42770-019-00049-7.

Alfaro-Chávez, A. L., J.-W. Liu, J. L. Porter, A. Goldman, and D. L. Ollis. 2019. Improving on nature's shortcomings: evolving a lipase for increased lipolytic activity, expression and thermostability. *Protein Engineering, Design & Selection: PEDS* 32 (1):13–24. doi: 10.1093/protein/gzz024.

Alizadeh, A. M., M. Masoomian, M. Shakooie, M. Z. Khajavi, and M. Farhoodi. 2022. Trends and applications of intelligent packaging in dairy products: A review. *Critical Reviews in Food Science and Nutrition* 62 (2):383–97. doi: 10.1080/10408398.2020.1817847.

Alkasir, R. S. J., M. Ornatska, and S. Andreescu. 2012. Colorimetric paper bioassay for the detection of phenolic compounds. *Analytical Chemistry* 84 (22):9729–37. doi: 10.1021/ac301110d.

Alnoch, R. C., A. A. Stefanello, Viviane, P. Martini, J. L. Richter, C. Mateo, E. M. de Souza, D. A. Mitchell, M. Muller-Santos, and N. Krieger. 2018. Co-expression, purification and characterization of the lipase and foldase of *Burkholderia* contaminans LTEB11. *International Journal of Biological Macromolecules* 116:1222–31. doi: 10.1016/j.ijbiomac.2018.05.086.

Alu'datt, M. H., T. Rababah, M. N. Alhamad, K. Ereifej, S. Gammoh, S. Kubow, and D. Tawalbeh. 2017. Preparation of mayonnaise from extracted plant protein isolates of chickpea, broad bean and lupin flour: chemical, physiochemical, nutritional and therapeutic properties. *Journal of Food Science and Technology* 54 (6):1395–405. doi: 10.1007/s13197-017-2551-6.

Alvarez, J. F., and V. J. Stella. 1989. The role of calcium ions and bile salts on the pancreatic lipase-catalyzed hydrolysis of triglyceride emulsions stabilized with lecithin. *Pharmaceutical Research* 6 (6):449–57. doi: 10.1023/a:1015956104500.

Alves-Bezerra, M., and D. E. Cohen. 2017. Triglyceride metabolism in the liver. *Comprehensive Physiology* 8 (1):1–22. doi: 10.1002/cphy.c170012.

Amit, K. S., M. M. Uddin, R. Rahman, S. M. R. Islam, and M. S. Khan. 2017. A review on mechanisms and commercial aspects of food preservation and processing. *Agriculture & Food Security* 6 (1):1–22. doi: 10.1186/s40066-017-0130-8.

Andriianova, N. A., Y. N. Biglova, and A. G. Mustafin. 2020. Effect of structural factors on the physicochemical properties of functionalized polyanilines. *RSC Advances* 10 (13):7468–91. doi: 10.1039/C9RA08644G.

Andualema, B., and A. Gessesse. 2012. Microbial lipases and their industrial applications: Review. *Biotechnology (Faisalabad)* 11 (3):100–18. doi: 10.3923/biotech.2012.100.118.

Anobom, C. D., A. S. Pinheiro, R. A. De-Andrade, E. C. G. Aguieiras, G. C. Andrade, M. V. Moura, R. V. Almeida, D., and M. Freire. 2014. From structure to catalysis: Recent developments in the biotechnological applications of lipases. *BioMed Research International* 2014:684506. Article ID 684506:doi.org/10.1155/2014/684506 doi: 10.1155/2014/684506.

Ansari, A. S., and Q. Husain. 2012. Potential applications of enzymes immobilized on/in nano materials: A review. *Biotechnology Advances* 30 (3):512–23. doi: 10.1016/j.biotechadv.2011.09.005.

Ansorena, D., M. J. Zapelena, I. Astiasarán, and J. Bello. 1998. Addition of palatase M (lipase from *Rhizomucor miehei*) to dry fermented sausages: Effect over lipolysis and study of the further oxidation process by GC MS. *Journal of Agricultural and Food Chemistry* 46 (8):3244–8. doi: 10.1021/jf980103p.

Anton, M. 2013. Egg yolk: Structures, functionalities and processes. *Journal of the Science of Food and Agriculture* 93 (12):2871–80. doi: 10.1002/jsfa.6247.

Arana-Peña, S., D. Carballares, Á. Berenguer-Murcia, A. R. Alcántara, R. C. Rodrigues, and R. Fernandez-Lafuente. 2020. One-pot use of combilipases for full modification of oils and fats: Multifunctional and heterogeneous substrates. *Catalysts* 10 (6):605. doi: 10.3390/catal10060605.

Arbige, M. V., and W. H. Pitcher. 1989. Industrial enzymology: A look towards the future. *Trends in Biotechnology* 7 (12):330–5. doi: 10.1016/0167-7799(89)90032-2.

Armand, M., B. Pasquier, M. André, P. Borel, M. Senft, J. Peyrot, J. Salducci, H. Portugal, V. Jaussan, and D. Lairon. 1999. Digestion and absorption of 2 fat emulsions with different droplet sizes in the human digestive tract. *The American Journal of Clinical Nutrition* 70 (6):1096–106. doi: 10.1093/ajcn/70.6.1096.

Augustyniak, W., A. A. Brzezinska, T. Pijning, H. Wienk, R. Boelens, B. W. Dijkstra, and M. T. Reetz. 2012. Biophysical characterization of mutants of *Bacillus subtilis* lipase evolved for thermostability: Factors contributing to increased activity retention. *Protein Science: A Publication of the Protein Society* 21 (4):487–97. doi: 10.1002/pro.2031.

Ayinla, Z. A., A. N. Ademakinwa, R. A. Gross, and F. K. Agboola. 2021. Biochemical and biophysical characterization of a small purified lipase from *Rhizopus oryzae* ZAC3. *Biocatalysis and Biotransformation* 39 (1):1–14. India: Wiley. doi: 10.1080/10242422.2021.1883006.

Badellino, K. O., M. L. Wolfe, M. P. Reilly, and D. J. Rader. 2005. Endothelial lipase concentrations are increased in metabolic syndrome and associated with coronary atherosclerosis. *PLoS Medicine* 3 (2):e22. Doi.org.10.1371/journal.pmed.0030022 doi: 10.1371/journal.pmed.0030022.

Baek, H. J., M.-J. Han, S. H. Lee, and S. Y. Lee. 2010. Enhanced display of lipase on the Escherichia coli cell surface, based on transcriptome analysis. *Applied and Environmental Microbiology* 76 (3):971–3. doi: 10.1128/AEM.02463-09.

Bakir, Z. B., and K. Metin. 2016. Purification and characterization of an alkali-thermostable lipase from thermophilic *Anoxybacillus flavithermus* HBB 134. *Journal of Microbiology and Biotechnology* 26 (6):1087–97. doi: 10.4014/jmb.1512.12056.

Balcão, V. M., A. L. Paiva, and F. X. Malcata. 1996. Bioreactors with immobilized lipases: State of the art. *Enzyme and Microbial Technology* 18 (6):392–416. doi: 10.1016/0141-0229(95)00125-5.

Baloch, M. K., and G. Hameed. 2005. Emulsification of oil in water as affected by different parameters. *Journal of Colloid and Interface Science* 285 (2):804–13. doi: 10.1016/j.jcis.2004.11.070.

Ban, K., S. Hama, K. Nishizuka, M. Kaieda, T. Matsumoto, A. Kondo, H. Noda, and H. Fukuda. 2002. Repeated use of whole-cell biocatalysts immobilized within biomass support particles for biodiesel fuel production. *Journal of Molecular Catalysis B: Enzymatic* 17 (3–5):157–65. doi: 10.1016/S1381-1177(02)00023-1.

Bancerz, R., G. Ginalska, J. Fiedurek, and A. Gromada. 2005. Cultivation conditions and properties of extracellular crude lipase from the psychrotrophic fungus *Penicillium chrysogenum* 9'. *Journal of Industrial Microbiology & Biotechnology* 32 (6):253–60. doi: 10.1007/s10295-005-0235-0.

Baneyx, F. 1999. Recombinant protein expression in *Escherichia coli*. *Current Opinion in Biotechnology* 10 (5):411–21. doi: 10.1016/S0958-1669(99)00003-8.

Barbosa, O., C. Ortiz, R. Torres, and R. Fernandez-Lafuente. 2011. Effect of the immobilization protocol on the properties of lipase B from *Candida antarctica* in organic media: Enantiospecifc production of atenolol acetate. *Journal of Molecular Catalysis B: Enzymatic* 71 (3–4):124–32. doi: 10.1016/j.molcatb.2011.04.008.

Barrett, D. M., J. C. Beaulieu, and R. Shewfelt. 2010. Color, flavor, texture, and nutritional quality of fresh-cut fruits and vegetables: Desirable levels, instrumental and sensory measurement, and the effects of processing. *Critical Reviews in Food Science and Nutrition* 50 (5):369–89. doi: 10.1080/10408391003626322.

Barros, M., L. F. Fleuri, and G. A. Macedo. 2010. Seed lipases: Sources, applications, and properties-a review. *Brazilian Journal of Chemical Engineering* 27 (1):15–29. doi: 10.1590/S0104-66322010000100002.

Basso, A., and S. Serban. 2019. Industrial applications of immobilized enzymes -A review. *Molecular Catalysis* 479 (2019):110607. doi: 10.1016/j.mcat.2019.110607.

Batumalaie, K., E. Khalili, N. A. Mahat, F. Z. Huyop, and R. A. Wahab. 2018. A statistical approach for optimizing the protocol for over-expressing lipase KV1 in *Escherichia coli*: Purification and characterization. *Biotechnology & Biotechnological Equipment* 32 (1):69–87. doi: 10.1080/13102818.2017.1407670.

Bayne, L., R. V. Ulijn, and P. J. Halling. 2013. Effect of pore size on the performance of immobilised enzymes. *Chemical Society Reviews* 42 (23):9000–10. doi: 10.1039/c3cs60270b.

Becker, P., I. Abu-Reesh, S. Markossian, G. Antranikian, and H. Märkl. 1997. Determination of the kinetic parameters during continuous cultivation of the lipase-producing thermophile *Bacillus* sp. IHI-91 on olive oil. *Applied Microbiology and Biotechnology* 48 (2):184–90. doi: 10.1007/s002530051036.

Bedran-Russo, A. K., G. F. Pauli, S.-N. Chen, J. McAlpine, C. S. Castellan, R. S. Phansalkar, T. R. Aguiar, C. M. P. Vidal, J. G. Napotilano, J.-W. Nam, et al. 2014. Dentin biomodification: Strategies, renewable resources and clinical applications. *Dental Materials: Official Publication of the Academy of Dental Materials* 30 (1):62–76. doi: 10.1016/j.dental.2013.10.012.

Beisson, F., A. Tiss, C. Rivière, and R. Verger. 2000. Methods for lipase detection and assay: A critical review. *European Journal of Lipid Science and Technology* 102 (2):133–53. doi: 0931-5985/2000/0202-0133 $17.50+.50/0 doi: 10.1002/(SICI)1438-9312(200002)102:2<133::AID-EJLT133>3.0.CO;2-X.

Benzinger, W., A. Becker, and K. J. Hüttinger. 1996. Chemistry and kinetics of chemical vapor deposition of pyrocarbon: I. Fundamentals of kinetics and chemical reaction engineering. *Carbon* 34 (8):957–66. doi: 10.1016/0008-6223(96)00010-3.

Bezerra, C. S., C. M. G. D. F. Lemos, M. D. Sousa, and L. R. B. Goncalves. 2015. Enzyme immobilization onto renewable polymeric matrixes: Past, present, and future trends. *Journal of Applied Polymer Science* 132(26):1–15. doi:10.1002/app.42125.

Bezerra, R. M., D. M. A. Neto, W. S. Galvão, N. S. Rios, A. C. L. d M Carvalho, M. A. Correa, F Bohn, R Fernandez-Lafuente, P. B. Fechine, M. C. De Mattos, et al. 2017. Design of a lipase-nano particle biocatalysts and its use in the kinetic resolution of medicament precursors. *Biochemical Engineering Journal* 132(26):104–15. doi:10.1016/j.bej.2017.05.024.

Bilal, M., M. Asgher, H. Cheng, Y. Yan, Hafiz, and M. N. Iqbal. 2019. Multi-point enzyme immobilization, surface chemistry, and novel platforms: A paradigm shift in biocatalyst design. *Critical Reviews in Biotechnology* 39 (2):202–19. doi: 10.1080/07388551.2018.1531822.

Bilal, M., C. D. Fernandes, T. Mehmood, F. Nadeem, Q. Tabassam, and L. F. R. Ferreira. 2021. Immobilized lipases-based nano-biocatalytic systems - A versatile platform with incredible biotechnological potential. *International Journal of Biological Macromolecules* 175 (1):108–22. doi: 10.1016/j.ijbiomac.2021.02.010.

Bjurlin, M. A., S. Bloomer, and M. J. Haas. 2001. Composition and activity of commercial triacylglycerol acylhydrolase preparations. *Journal of the American Oil Chemists' Society* 78 (2):153–60. doi: 10.1007/s11746-001-0236-9.

Blank, K., J. Morfill, H. Gumpp, and H. E. Gaub. 2006. Functional expression of *Candida antarctica* lipase B in Eschericha coli. *Journal of Biotechnology* 125 (4):474–83. doi: 10.1016/j.jbiotec.2006.04.004.

Bloomer, S., P. Adlercreutz, and B. Mattiasson. 1990. Triglyceride interesterification by lipases. 1. Cocoa butter equivalents from a fraction of palm oil. *Journal of the American Oil Chemists' Society* 67 (8):519–24. 519-524. doi: 10.1007/BF02540759.

Bocchetta, P. 2020. Ionotropic gelation of chitosan for next-generation composite proton. Conducting. Flat structures. *Molecules* 25 (7):1632. doi: 10.3390/molecules25071632.

Bockelmann, W. 2000. Secondary cheese starter cultures. *Technology of Cheesemaking*: ed. B. A. Law, A. Y. Tamime, 193–230. USA: Blackwell Publishing Ltd. doi: 10.1002/9781444323740.

Boel, E., B. Huge-Jensen, M. Christensen, L. Thim, and N. P. Fiil. 1988. *Rhizomucor miehei* triglyceride lipase is synthesized as a precursor. *Lipids* 23 (7):701–6. doi: 10.1007/BF02535672.

Boller, T., C. Meier, and S. Menzler. 2002. Eupergit oxirane acrylic beads: How to make enzymes fit for biocatalysis. *Organic Process Research & Development* 6 (4):509–19. doi: 10.1021/op015506w.

Bora, L., D. Gohain, and R. Das. 2013. Recent advances in production and biotechnological applications of thermostable and alkaline bacterial lipases. *Journal of Chemical Technology & Biotechnology* 88 (11):n/a–1970. doi: 10.1002/jctb.4170.

Borkar, P. S., R. G. Bodade, S. R. Rao, and C. N. Khobragade. 2009. Purification and characterization of extracellular lipase from a new strain: *Pseudomonas aeruginosa* SRT 9. *Brazilian Journal of Microbiology: [Publication of the Brazilian Society for Microbiology]* 40 (2):358–66. doi: 10.1590/S1517-83822009000200028.

Borrelli, G. M., and D. Trono. 2015. Recombinant lipases and phospholipases and their use as biocatalysts for industrial applications. *International Journal of Molecular Sciences* 16 (9):20774–840. doi: 10.3390/ijms160920774.

Bovara, R., G. Carrea, G. Ottolina, and S. Riva. 1993. Water activity does not influence the enantioselectivity of lipase PS and lipoprotein lipase in organic solvents. *Biotechnology Letters* 15 (2):169–74. doi: 10.1007/BF00133018.

Bracco, U. 1994. Effect of triglyceride structure on fat absorption. *The American Journal of Clinical Nutrition* 60 (6 Suppl):1002S–9S. doi: 10.1093/ajcn/60.6.1002S.

Brena, B., P. González-Pombo, and F. Batista-Viera. 2013. Immobilization of enzymes: A literature survey. In *Immobilization of enzymes and cells. Methods in molecular biology (methods and protocols)*, ed. J. Guisan, vol 1051. Totowa, NJ: Humana Press. doi: 10.1007/978-1-62703-550-7_2.

Brennan, N. M., T. M. Cogan, M. Loessner, and S. Scherer. 2004. Bacterial surface-ripened cheeses. *Cheese: chemistry, Physics and Microbiology* 2:199–225. doi: 10.1016/S1874-558X.(04)80045-9

Brindley, D. N. 1991. Metabolism of triacylglycerols. In *New comprehensive biochemistry*, ed. D. E. Vance, J. E. Vance. vol. 20:171–203. London: Elsevier. doi: 10.1016/S0167-7306(08)60334-8.

Brocca, S., F. Secundo, M. Ossola, L. Alberghina, G. Carrea, and M. Lotti. 2003. Sequence of the lid affects activity and specificity of Candida rugosa lipase isoenzymes. *Protein Science: A Publication of the Protein Society* 12 (10):2312–9. doi: 10.1110/ps.0304003.

Budhwani, A. A., Aziz, A. Maqbool, T. Hussain, and M. N. Syed. 2019. Production of biodiesel by enzymatic transesterification of non-edible *Salvadora persica* (Pilu) oil and crude coconut oil in a solvent-free system. *Bioresources and Bioprocessing* 6 (1):1–9. doi: 10.1186/s40643-019-0275-3.

Bueso, F., L. Moreno, M. Cedeño, and K. Manzanarez. 2015. Lipase-catalyzed biodiesel production and quality with *Jatropha curcas* oil: Exploring its potential for Central America. *Journal of Biological Engineering* 9 (1):1–7. doi: 10.1186/s13036-015-0009-9.

Califano, V., and A. Costantini. 2020. Immobilization of cellulolytic enzymes in mesostructured silica materials. *Catalysts* 10 (6):706. doi: 10.3390/catal10060706.

Carneiro, E., A. K. P. Araújo, Bastos, U. M. F. de Oliveira, L. J. B. L. de Matos, W. S. Adriano, R. R. d. C. Monteiro, J. C. S. d. Santos, and L. R. B. Gonçalves. 2020. Improving the catalytic features of the lipase from *Rhizomucor miehei* immobilized on chitosan-based hybrid matrices by altering the chemical activation conditions. *Química Nova* 43 (9):1234–9. doi: 10.21577/0100-4042.20170615.

Carrie, A. T., P. J. Delaquis, and G. Mazza. 1999. Detection and measurement of microbial lipase activity: A review. *Critical Reviews in Food Science and Nutrition* 39 (2):165–87. doi: 10.1080/10408399908500492.

Carvalho, N. B., J. M. P. Barbosa, M. V. S. Oliveira, A. T. Fricks, Á. S. Lima, and C. M. F. Soares. 2013. Biochemical properties of *Bacillus* sp. ITP-001 lipase immobilized with a sol gel process. *Química Nova* 36 (1):52–8. doi: 10.1590/S0100-40422013000100010.

Carvalho, P. d. O., F. J. Contesini, R. Bizaco, S. Calafatti, and G. A. Macedo. 2006. Optimization of enantioselective resolution of racemic ibuprofen by native lipase from *Aspergillus niger*. *Journal of Industrial Microbiology & Biotechnology* 33 (8):713–8. doi: 10.1007/s10295-006-0138-8.

Casas-Godoy, L., S. Duquesne, F. Bordes, G. Sandoval, and A. Marty. 2012. Lipase: An overview. Lipases and phospholipases SE-1. *Methods in Molecular Biology (Methods and Protocols)*, ed. G. Sandoval, vol. 861, pp. 3–30. New York, NY: Humana Press. doi: 10.1007/978-1-61779-600-5_1

Chaffin, W. L. 2008. *Candida albicans* cell wall proteins. *Microbiology and Molecular Biology Reviews: MMBR* 72 (3):495–544. doi: 10.1128/MMBR.00032-07.

Chahinian, H., and L. Sarda. 2009. Distinction between esterases and lipases: Comparative biochemical properties of sequence-related carboxylesterases. *Protein and Peptide Letters* 16 (10):1149–61. doi: 10.2174/092986609789071333.

Chamorro, S., J. M. Sanchez-Montero, A. R. Alcantara, and J. V. Sinisterra. 1998. Treatment of *Candida rugosa* lipase with short-chain polar organic solvents enhances its hydrolytic and synthetic activities. *Biotechnology Letters* 20 (5):499–505. doi: 10.1023/A:1005448431237.

Chandra, P., Enespa, and D. P. Singh. 2020a. Microplastic degradation by bacteria in the aquatic ecosystem. In *Microorganisms for sustainable environment and health*, 431–467. India: Elsevier. doi. org/10.1016/B978-0-12-819001-2.00022-X

Chandra, P., Enespa, R. Singh, and P. K. Arora. 2020b. Microbial lipases and their industrial applications: A comprehensive review. *Microbial Cell Factories* 19 (1):1–42. doi: 10.1186/s12934-020-01428-8.

Chandra, P., and D. P. Singh. 2014. Removal of Cr (VI) by a halotolerant bacterium *Halomonas* sp. CSB 5 isolated from sāmbhar salt Lake Rajasthan (India). *Cell Molecular Biology* 60 (5):64–72. PMID: 25535715.

Chang, H.-J., and J.-H. Lee. 2021. Regiospecific positioning of palmitic acid in triacylglycerol structure of enzymatically modified lipids affects physicochemical and in vitro digestion properties. *Molecules* 26 (13):4015. doi: 10.3390/molecules26134015.

Chapman, J., A. E. Ismail, and C. Z. Dinu. 2018. Industrial applications of enzymes: Recent advances, techniques, and outlooks. *Catalysts* 8 (6):238. doi: 10.3390/catal8060238.

Chen, F., J. Sun, Z. Han, X. Yang, J. Xian, A. Lv, X. Hu, and H. Shi. 2019. Isolation, identification and characteristics of Aeromonas veronii from diseased crucian carp (Carassius auratus gibelio). *Frontiers in Microbiology* 10:2742. doi: 10.3389/fmicb.2019.02742.

Cheng, Y., B. Ma, C.-P. Tan, O.-M. Lai, W. Panpipat, L.-Z. Cheong, and C. Shen. 2020. Hierarchical macro-microporous ZIF-8 nanostructures as efficient nano-lipase carriers for rapid and direct electrochemical detection of nitrogenous diphenyl ether pesticides. *Sensors and Actuators B: Chemical* 321 (128477):128477. doi: 10.1016/j.snb.2020.128477.

Chin, V. K., T. Y. Lee, B. Rusliza, and P. P. Chong. 2016. Dissecting *Candida albicans* infection from the perspective of *C. albicans* virulence and omics approaches on host-pathogen interaction: A review. *International Journal of Molecular Sciences* 17 (10):1643. doi: 10.3390/ijms17101643.

Cho, Y. K., and J. E. Bailey. 1977. Enzyme immobilization on activated carbon: Alleviation of enzyme deactivation by hydrogen peroxide. *Biotechnology and Bioengineering* 19 (5):769–75. doi: 10.1002/bit.260190514.

Choi, J. H., and S. Y. Lee. 2004. Secretory and extracellular production of recombinant proteins using *Escherichia coli*. *Applied Microbiology and Biotechnology* 64 (5):625–35. doi: 10.1007/s00253-004-1559-9.

Coelho, A. L., Silva, and R. C. Orlandelli. 2021. Immobilized microbial lipases in the food industry: A systematic literature review. *Critical Reviews in Food Science and Nutrition* 61 (10):1689–703. doi: 10.1080/10408398.2020.1764489.

Colón-Ramos, U., R. Monge-Rojas, and H. Campos. 2014. Impact of WHO recommendations to eliminate industrial trans-fatty acids from the food supply in Latin America and the Caribbean. *Health Policy and Planning* 29 (5):529–41. doi: 10.1093/heapol/czt034.

Contesini, F. J., M. G. Davanço, G. P. Borin, K. G. Vanegas, J. P. Gonçalves Cirino, R. R. de Melo, U. H. Mortensen, K. Hildén, D. R. Campos, P. de, et al. 2020. Advances in recombinant lipases: Production, engineering, immobilization, and application in the

pharmaceutical industry. *Catalysts* 10 (9):1032. doi: 10.3390/catal10091032.
Contesini, F. J., D. B. Lopes, G. A. Macedo, M. da, G. Nascimento, P. de, and O. Carvalho. 2010. *Aspergillus* sp. lipase: Potential biocatalyst for industrial use. *Journal of Molecular Catalysis B: Enzymatic* 67 (3–4):163–71. doi: 10.1016/j.molcatb.2010.07.021.
Cooney, M. J. 2017. Kinetic measurements for enzyme immobilization. In *Enzyme stabilization and immobilization. Methods in molecular biology (methods and protocols)*, ed. S. Minteer, vol. 679. Totowa, NJ: Humana Press. doi: 10.1007/978-1-60761-895-9_17.
Corrêa, R. C. G., R. M. Peralta, A. Bracht, Isabel, and C. F. R. Ferreira. 2017. The emerging use of mycosterols in the food industry along with the current trend of extended use of bioactive phytosterols. *Trends in Food Science & Technology* 67:19–35. doi: 10.1016/j.tifs.2017.06.012.
Cregg, J. M., J. L. Cereghino, J. Shi, and D. R. Higgins. 2000. Recombinant protein expression in *Pichia pastoris*. *Molecular Biotechnology* 16 (1):23–52. doi: 10.1385/MB:16:1:23.
Crielly, E. M., N. A. Logan, and A. Anderton. 1994. Studies on the Bacillus flora of milk and milk products. *The Journal of Applied Bacteriology* 77 (3):256–63. doi: 10.1111/j.1365-2672.1994.tb03072.x.
da Silva, V. C., Fernandes, F. J. Contesini, and P. d O. Carvalho. 2009. Enantioselective behavior of lipases from *Aspergillus niger* immobilized in different supports. *Journal of Industrial Microbiology & Biotechnology* 36 (7):949–54. doi: 10.1007/s10295-009-0573-4.
Dalgleish, D. G. 1993. The enzymatic coagulation of milk. In *Cheese: Chemistry, physics, and microbiology*, ed. P. F. Fox, 69–100. Boston, MA: Springer. doi: 10.1007/978-1-4615-2650-6_3.
Dandavate, V., H. Keharia, and D. Madamwar. 2011. Ester synthesis using *Candida rugosa* lipase immobilized on magnetic nanoparticles. *Biocatalysis and Biotransformation* 29 (1):37–45. doi: 10.3109/10242422.2010.550044.
Darvishi, F. 2012. Expression of native and mutant extracellular lipases fromYarrowia lipolytica in Saccharomyces cerevisiae. *Microbial Biotechnology* 5 (5):634–41. doi: 10.1111/j.1751-7915.2012.00354.x.
Datta, S., L. R. Christena, and Y. R. S. Rajaram. 2013. Enzyme immobilization: An overview on techniques and support materials. *3 Biotech* 3 (1):1–9. doi: 10.1007/s13205-012-0071-7.
Davis, B. A., A. Nagarajan, L. R. Forrest, and S. K. Singh. 2016. Mechanism of paroxetine (paxil) inhibition of the serotonin transporter. *Scientific Reports* 6 (1):23789–13. doi: 10.1038/srep23789.
De Jong, E. D., W. J. H V. A. N. Berkel, R. P. V. A. N D. E. R. Zwan, and J. A. M D. E. Bont. 1992. Purification and characterization of vanillyl-alcohol oxidase from Penicillium simplicissimum. A novel aromatic alcohol oxidase containing covalently bound FAD. *European Journal of Biochemistry* 208 (3):651–7. doi: 10.1111/j.1432-1033.1992.tb17231.x.
de Marco, A. 2009. Strategies for successful recombinant expression of disulfide bond-dependent proteins in *Escherichia coli*. *Microbial Cell Factories* 8 (26):26–14. doi: 10.1186/1475-2859-8-26.
De Maria, L., J. Vind, K. M. Oxenbøll, A. Svendsen, and S. Patkar. 2007. Phospholipases and their industrial applications. *Applied Microbiology and Biotechnology* 74 (2):290–300. doi: 10.1007/s00253-006-0775-x.
de Moura Barboza, A., A. B. d. Silva, E. M. d. Silva, W. P. d. Souza, M. A. Soares, L. G. d. Vasconcelos, A. J. Terezo, and M. Castilho. 2019. A biosensor based on microbial lipase immobilized on lamellar zinc hydroxide-decorated gold nanoparticles for carbendazim determination. *Analytical Methods* 11 (42):5388–97. doi: 10.1039/C9AY01600G.
de Souza, S. P., R. A. D d. Almeida, G. G. Garcia, R. A. C. Leão, J. Bassut, R. O. M. A d. Souza, and I. Itabaiana. Jr. 2018. Immobilization of lipase B from *Candida antarctica* on epoxy-functionalized silica: Characterization and improving biocatalytic parameters. *Journal of Chemical Technology & Biotechnology* 93 (1):105–11. doi: 10.1002/jctb.5327.
de Souza, T. C., T. d S. Fonseca, J. de Sousa Silva, P. J. M. Lima, C. A. C. G. Neto, R. R. C. Monteiro, M. V. P. Rocha, M. C. de Mattos, J. C. S. Dos Santos, and L. R. B. Gonçalves. 2020. Modulation of lipase B from *Candida antarctica* properties via covalent immobilization on eco-friendly support for enzymatic kinetic resolution of rac-indanyl acetate. *Bioprocess and Biosystems Engineering* 43 (12):2253–68. doi: 10.1007/s00449-020-02411-8.
Delorme, V., R. Dhouib, S. Canaan, F. Fotiadu, F. Carrière, and J. F. Cavalier. 2011. Effects of surfactants on lipase structure, activity, and inhibition. *Pharmaceutical Research* 28 (8):1831–42. doi: 10.1007/s11095-010-0362-9.
Demuner, B. J., Nei, P. Junior, and A. Antunes. 2011. Technology prospecting on enzymes for the pulp and paper industry. *Journal of Technology Management & Innovation* 6 (3):148–58. doi: 10.4067/S0718-27242011000300011.
DiCosimo, R., J. McAuliffe, A. J. Poulose, and G. Bohlmann. 2013. Industrial use of immobilized enzymes. *Chemical Society Reviews* 42 (15):6437–74. doi: 10.1039/c3cs35506c.
Dinanta, U., Q. Azis Boing Sitanggang, D. R. Adawiyah, and P. Hariyadi. 2019. Lipase-catalyzed interesterification for the synthesis of medium-long-medium (MLM) structured Lipids - A Review. *Food Technology and Biotechnology* 57 (3):305–18. doi: 10.17113/ftb.57.03.19.6025.
Dobreva, V., B. Zhekova, and G. Dobrev. 2019. Use of aqueous two-phase and three-phase partitioning systems for purification of lipase obtained in solid-state fermentation by *Rhizopus arrhizus*. 2019. *The Open Biotechnology Journal* 13 (1):27–36. doi: 10.2174/1874070701913010027.
Doolittle, M. H., and M. Péterfy. 2010. Mechanisms of lipase maturation. *Clinical Lipidology* 5 (1):117–30. doi: 10.2217/clp.09.84.
Dugat-Bony, E., C. Straub, A. Teissandier, D. Onésime, V. Loux, C. Monnet, F. Irlinger, S. Landaud, M.-N. Leclercq-Perlat, P. Bento, et al. 2015. Overview of a surface-ripened cheese community functioning by meta-omics analyses. *PloS One* 10 (4):e0124360. doi. org./101371journal pone 0124360 doi: 10.1371/journal.pone.0124360.
Dugourd, D., C. Martin, C. R. Rioux, M. Jacques, and J. Harel. 1999. Characterization of a periplasmic ATP-binding cassette iron import system of *Brachyspira* (Serpulina) hyodysenteriae. *Journal of Bacteriology* 181 (22):6948–57. doi: 10.1128/JB.181.22.6948-6957.1999.
Edwards, F. L., and P. B. Tchounwou. 2005. Environmental toxicology and health effects associated with methyl parathion exposure-a scientific review. *International Journal of Environmental Research and Public Health* 2 (3–4):430–41. doi: 10.3390/ijerph2005030007.
Eggert, T., C. Leggewie, M. Puls, W. Streit, G. Van Pouderoyen, B. W. Dijkstra, and K.-E. Jaeger. 2004. Novel biocatalysts by identification and design. *Biocatalysis and Biotransformation* 22 (2):141–6. doi: 10.1080/10242420410001710056.
Eissa, S., C. Tlili, L. L'Hocine, and M. Zourob. 2012. Electrochemical immunosensor for the milk allergen β-lactoglobulin based on electrografting of organic film on graphene modified screen-printed carbon electrodes. *Biosensors & Bioelectronics* 38 (1):308–13. doi: 10. 1016/j.bios.2012.06.008.
El Khattabi, M., P. V. Gelder, W. Bitter, and J. Tommassen. 2003. Role of the calcium ion and the disulfide bond in the *Burkholderia glumae* lipase. *Journal of Molecular Catalysis B: Enzymatic* 22 (5–6):329–38. doi.org/10.1016/S1381-1177 (03)00047-X doi: 10.1016/S1381-1177(03)00047-X.
El Seoud, O. A., W. J. Baader, and E. L. Bastos. 2016. Practical chemical kinetics in solution. *Encyclopedia of Physical Organic Chemistry* American Cancer Society, eds. Z. Wang, U. Wille, E. Juaristi, 1–68. Hoboken, New Jersey: John Wiley & Sons. doi: 10.1002/9781118468586.epoc1012.
Ene-Obong, H. N., and E. Carnovale. 1992. Nigerian soup condiments: Traditional processing and potential as dietary fiber sources. *Food Chemistry* 43 (1):29–34. doi.org/10.1016/0308-8146 (92)90237-V doi: 10.1016/0308-8146(92)90237-V.
Engasser, J.-M., and C. S. A. B. A. Horvath. 1975. Electrostatic effects on the kinetics of bound enzymes. *The Biochemical Journal* 145 (3):431–5. doi: 10.1042/bj1450431.
Eom, G., J. Tae, K. Song, J. H. Ahn, Y. S. Seo, and J. S. Rhee. 2005. Enhancement of the efficiency of secretion of heterologous lipase in *Escherichia coli* by directed evolution of the ABC transporter system. *Applied and Environmental Microbiology* 71 (7):3468–74. doi: 10.1128/AEM.71.7.3468-3474.2005.

Ewens, H., L. Metilli, and E. Simone. 2021. Analysis of the effect of recent reformulation strategies on the crystallization behaviour of cocoa butter and the structural properties of chocolate. *Current Research in Food Science* 4:105–14. doi: 10.1016/j.crfs.2021.02.009.

Facin, B. R., M. S. Melchiors, A. Valério, J. V. Oliveira, and D. d. Oliveira. 2019. Driving immobilized lipases as biocatalysts: 10 years state of the art and future prospects. *Industrial & Engineering Chemistry Research* 58 (14):5358–78. doi: 10.1021/acs.iecr.9b00448.

Falcocchio, S., C. Ruiz, F. I J. Pastor, L. Saso, and P. Diaz. 2006. *Propionibacterium acnes* GehA lipase, an enzyme involved in acne development, can be successfully inhibited by defined natural substances. *Journal of Molecular Catalysis B: Enzymatic* 40 (3–4):132–7. doi: 10.1016/j.molcatb.2006.02.011.

Farag, A. M., and M. A. Hassan. 2004. Purification, characterization, and immobilization of a keratinase from *Aspergillus oryzae*. *Enzyme and Microbial Technology* 34 (2):85–93. doi: 10.1016/j.enzmictec.2003.09.002.

Faraldos, J. A., J.-L. Giner, D. H. Smith, M. Wilson, K. Ronhovde, E. Wilson, David Clevette, A. E. Holmes, and K. Rouhier. 2011. Enzymatic resolution of 1-phenylethanol and formation of a diastereomer: An undergraduate H NMR experiment to introduce chiral chemistry. *Journal of Chemical Education* 88 (3):334–6. doi: 10.1021/ed100325p.

Farfán, M., A. Álvarez, A. Gárate, and P. Bouchon. 2015. Comparison of chemical and enzymatic interesterification of fully hydrogenated soybean oil and walnut oil to produce a fat base with adequate nutritional and physical characteristics. *Food Technology and Biotechnology* 53 (3):361–6. doi: 10.17113/ftb.53.03.15.3854.

Farrokh, P., B. Yakhchali, and A. A. Karkhane. 2014. Cloning and characterization of newly isolated lipase from *Enterobacter* sp. Bn12. *Brazilian Journal of Microbiology: [Publication of the Brazilian Society for Microbiology]* 45 (2):677–87. doi: 10.1590/s1517-83822014000200042.

Fernandez, I., and N. Khiar. 2003. Recent developments in the synthesis and utilization of chiral sulfoxides. *Chemical Reviews* 103 (9):3651–706. doi: 10.1021/cr990372u.

Fernandez-Lafuente, R. 2009. Stabilization of multimeric enzymes: Strategies to prevent subunit dissociation. *Enzyme and Microbial Technology* 45 (6–7):405–18. doi: 10.1016/j.enzmictec.2009.08.009.

Fernandez-Lorente, G., J. Rocha-Martín, and J. M. Guisan. 2020. Immobilization of lipases by adsorption on hydrophobic supports: Modulation of enzyme properties in biotransformations in anhydrous media. In *Immobilization of enzymes and cells*, 143–58. New York, NY: Humana. doi: 10.1007/978-1-0716-0215-7_9.

Ferreira-Dias, S., G. Sandoval, F. Plou, and F. Valero. 2013. The potential use of lipases in the production of fatty acid derivatives for the food and nutraceutical industries. *Electronic Journal of Biotechnology* 16 (3):12. doi: 10.2225/vol16-issue3-full.text-5

Fickers, P., J. Destain, and P. Thonart. 2009. Improvement of *Yarrowia lipolytica* lipase production by fed-batch fermentation. *Journal of Basic Microbiology* 49 (2):212–5. doi: 10.1002/jobm.200800186.

Foegeding, E. A., P. J. Luck, and J. P. Davis. 2006. Factors determining the physical properties of protein foams. *Food Hydrocolloids* 20 (2–3):284–92. doi: 10.1016/j.foodhyd.2005.03.014.

Forde, A., and G. F. Fitzgerald. 2000. Biotechnological approaches to the understanding and improvement of mature cheese flavor. *Current Opinion in Biotechnology* 11 (5):484–9. doi: 10.1016/S0958-1669.(00)00130-0

Fourie, R., O. O. Kuloyo, B. M. Mochochoko, J. Albertyn, and C. H. Pohl. 2018. Iron at the Centre of Candida albicans Interactions. *Frontiers in Cellular and Infection Microbiology* 8 (2018):185. doi: 10.3389/fcimb.2018.00185.

Fox, P. F. 1993. Exogenous enzymes in dairy technology—a review 1. *Journal of Food Biochemistry* 17 (3):173–99. doi: 10.1111/j.1745-4514.1993.tb00466.x.

Frayn, K. N., F. Karpe, B. A. Fielding, I. A. Macdonald, and S. W. Coppack. 2003. Integrative physiology of human adipose tissue. *International Journal of Obesity* 27 (8):875–88. doi:10.1038/sj.ijo.0802326.

Fuglsang, C., Crone, C. Johansen, S. Christgau, and J. Adler-Nissen. 1995. Antimicrobial enzymes: Applications and future potential in the food industry. *Trends in Food Science & Technology* 6 (12):390–6. doi: 10.1016/S0924-2244(00)89217-1.

Fukunaga, K., N. Maruoka, Y. Sugimura, K. Nakao, and T. Shimizu. 1998. Preparation of the gemini detergent-lipase complexes and their high enzymatic activities in the transesterifications in homogeneous organic solvents. *Biotechnology Letters* 20 (12):1161–5. [Mismatch] doi: 10.1023/A:1005336705912.

Gácser, A., F. Stehr, C. Kröger, L. Kredics, W. Schäfer, and J. D. Nosanchuk. 2007. Lipase 8 affects the pathogenesis of *Candida albicans*. *Infection and Immunity* 75 (10):4710–8. doi: 10.1128/IAI.00372-07.

Galvão, W. S., B. B. Pinheiro, L. R. B. Golçalves, M. C. de Mattos, T. S. Fonseca, T. Regis, D. Zampieri, J. C. S. dos Santos, L. S. Costa, M. A. Correa, et al. 2018. Novel nanohybrid biocatalyst: Application in the kinetic resolution of secondary alcohols. *Journal of Materials Science* 53 (20):14121–37. doi: 10.1007/s10853-018-2641-5.

García, C., P. Hoyos, and M. J. Hernáiz. 2018. Enzymatic synthesis of carbohydrates and glycoconjugates using lipases and glycosidases in green solvents. *Biocatalysis and Biotransformation* 36 (2):131–40. doi: 10.1080/10242422.2017.1349760.

Garcia-Galan, C., Á. Berenguer-Murcia, R. Fernandez-Lafuente, and R. C. Rodrigues. 2011. Potential of different enzyme immobilization strategies to improve enzyme performance. *Advanced Synthesis & Catalysis* 353 (16):2885–904. doi: 10.1002/adsc.201100534.

Gargova, S., M. Sariyska, A. Angelov, and I. Stoilova. 2006. *Aspergillus niger* pH 2.1 optimum acid phosphatase with high affinity for phytate. *Folia Microbiologica* 51 (6):541–5. doi: 10.1007/BF02931618.

Gasmi, N., A. Ayed, N. Jean-Marc, and H. Kallel. 2011. Design of an efficient medium for heterologous protein production in *Yarrowia lipolytica*: case of human interferon alpha 2b. *Microbial Cell Factories* 10 (1):38–13. doi: 10.1186/1475-2859-10-38.

Gawas, S. D., S. V. Jadhav, and V. K. Rathod. 2016. Solvent free lipase catalysed synthesis of ethyl laurate: Optimization and kinetic studies . *Applied Biochemistry and Biotechnology* 180 (7):1428–45. doi: 10.1007/s12010-016-2177-6.

Gerits, L. R., B. Pareyt, K. Decamps, and J. A. Delcour. 2014. Lipases and their functionality in the production of wheat-based food systems. *Comprehensive Reviews in Food Science and Food Safety* 13 (5):978–89. doi: 10.1111/1541-4337.12085.

Ghosh, M., S. Avery, D. K. Bhattacharyya, and M. Ghosh. 2016. Preparation of human milk fat analogue by enzymatic interesterification reaction using palm stearin and fish oil. *Journal of Food Science and Technology* 53 (4):2017–24. doi: 10.1007/s13197-016-2180-5.

Gibbons, S. 2012. 'Legal highs'-novel and emerging psychoactive drugs: a chemical overview for the toxicologist. *Clinical Toxicology (Philadelphia, PA)* 50 (1):15–24. doi: 10.3109/15563650.2011.645952.

Gilbert, E. J., A. Cornish, and C. W. Jones. 1991. Purification and properties of extracellular lipase from *Pseudomonas aeruginosa* EF2. *Journal of General Microbiology* 137 (9):2223–9. doi: 10.1099/00221287-137-9-2223.

Golding, M., T. J. Wooster, L. Day, M. Xu, L. Lundin, J. Keogh, and P. Clifton. 2011. Impact of gastric structuring on the lipolysis of emulsified lipids. *Soft Matter* 7 (7):3513–23. doi: 10.1039/c0sm01227k.

Gonçalves, F. D., A. G. Silva, and C. Z. Guidini. 2019. Lipases: Sources, immobilization methods, and industrial applications. *Applied Microbiology and Biotechnology* 103 (18):7399–423. doi: 10.1007/s00253-019-10027-6.

González, T., H. N. M'Barek, A. E. Gomaa, H. Hajjaj, C. Zhen, and L. Dehua. 2019. Molecular cloning and expression of *Candida antarctica* lipase B in *Corynebacterium* genus. *Microbiology and Biotechnology Letters* 47 (4):546–54. doi: 10.4014/mbl.1905.05003.

Gonzalez-Baró, M. R., T. M. Lewin, and R. A. Coleman. 2007. Regulation of triglyceride metabolism II. Function of mitochondrial GPAT1 in the regulation of triacylglycerol biosynthesis and insulin action. *American Journal of Physiology. Gastrointestinal and Liver Physiology* 292 (5):G1195–G1199. doi: 10.1152/ajpgi.00553.2006.

Goodson, A., H. Robin, W. Summerfield, and I. Cooper. 2004. Migration of bisphenol A from can coatings-effects of damage, storage conditions and heating. *Food Additives and Contaminants* 21 (10):1015–26. doi: 10.1080/02652030400011387.

Gopinath, S. C. B., A. Hilda, T. Lakshmi Priya, G. Annadurai, and P. Anbu. 2003. Purification of lipase from *Geotrichum candidum*: Conditions optimized for enzyme production using Box–Behnken design. *World Journal of Microbiology and Biotechnology* 19 (7):681–9. doi: 10.1023/A:1025119222925.

Grönke, S., A. Mildner, S. Fellert, N. Tennagels, S. Petry, G. Müller, H. Jäckle, and R. P. Kühnlein. 2005. Brummer lipase is an evolutionary conserved fat storage regulator in *Drosophila*. *Cell Metabolism* 1 (5):323–30. doi: 10.1016/j.cmet.2005.04.003.]

Guerrand, D. 2017. Lipase's industrial applications: Focus on food and agroindustries. *OCL* 24 (4):D403. doi: 10.1051/ocl/2017031.

Guncheva, M., and D. Zhiryakova. 2011. Catalytic properties and potential applications of *Bacillus lipases*. *Journal of Molecular Catalysis B: Enzymatic* 68 (1):1–21. doi: 10.1016/j.molcatb.2010.09.002.

Gupta, R., N. Gupta, and P. Rathi. 2004. Bacterial lipases: An overview of production, purification and biochemical properties. *Applied Microbiology and Biotechnology* 64 (6):763–81. doi: 10.1007/s00253-004-1568-8.

Gutiérrez, A., J. C. d. Río, and A. T. Martínez. 2009. Microbial and enzymatic control of pitch in the pulp and paper industry. *Applied Microbiology and Biotechnology* 82 (6):1005–18. doi: 10.1007/s00253-009-1905-z.

Guzik, U., K. Hupert-Kocurek, and D. Wojcieszyńska. 2014. Immobilization as a strategy for improving enzyme properties-application to oxidoreductases. *Molecules (Basel, Switzerland)* 19 (7):8995–9018. doi: 10.3390/molecules19078995.

Handayani, N., D. Wahyuningrum, M. A. Zulfikar, S. Nurbaiti, and C. L. Radiman. 2016. The synthesis of biodiesel catalyzed by *Mucor miehei* lipase immobilized onto aminated polyethersulfone membranes. *Bioresources and Bioprocessing* 3 (1):1–11. doi: 10.1186/s40643-016-0098-4.

Haraldsson, G. G., P. A. Höskuldsson, S. T. Sigurdsson, F. Thorsteinsson, and S. Gudbjarnason. 1989. The preparation of triglycerides highly enriched with ω-3 polyunsaturated fatty acids via lipase-catalyzed interesterification. *Tetrahedron Letters* 30 (13):1671–4. doi.org/(00)99550-9 doi: 10.1016/S0040-4039.

Harini, T., J. Muddagoni, G. Sheelu, H. B. Rode, and T. Kumaraguru. 2021. Polymer supported cross-linked enzyme aggregates (CLEAs) of lipase B from *Candida antarctica*: An efficient and recyclable biocatalyst for reactions in both aqueous and organic media. *Biocatalysis and Biotransformation* 39 (1): 1–13. doi: 10.1080/10242422.2021.1885381.

Hartmann, M., and X. Kostrov. 2013. Immobilization of enzymes on porous silicas-benefits and challenges. *Chemical Society Reviews* 42 (15):6277–89. doi: 10.1039/c3cs60021a.

Hasan, F., A. A. Shah, and A. Hameed. 2006. Industrial applications of microbial lipases. *Enzyme and Microbial Technology* 39 (2):235–51. doi: 10.1016/j.enzmictec.2005.10.016.

Hasan, F., A. A. Shah, and A. Hameed. 2009. Methods for detection and characterization of lipases: A comprehensive review. *Biotechnology Advances* 27 (6):782–98. doi: 10.1016/j.biotechadv.2009.06.001.

Hazem, Z. M. 2009. Acute biliary pancreatitis: Diagnosis and treatment. *Saudi Journal of Gastroenterology: Official Journal of the Saudi Gastroenterology Association* 15 (3):147–55. doi: 10.4103/1319-3767.54740.

He, W.-S., H. Zhu, and Z.-Y. Chen. 2018. Plant sterols: Chemical and enzymatic structural modifications and effects on their cholesterol-lowering activity. *Journal of Agricultural and Food Chemistry* 66 (12):3047–62. doi: 10.1021/acs.jafc.8b00059.

Hellwig, S., J. Drossard, R. M. Twyman, and R. Fischer. 2004. Plant cell cultures for the production of recombinant proteins. *Nature Biotechnology* 22 (11):1415–22. doi: 10.1038/nbt1027.

Hemlata, B., Z. Uzma, and K. Tukaram. 2016. Substrate kinetics of thiol activated hyperthermostable alkaline lipase of *Bacillus sonorensis* 4R and its application in the bio-detergent formulation. *Biocatalysis and Agricultural Biotechnology* 8:104–11. doi: 10.1016/j.bcab.2016.08.008.

Henry, J. 2009. Processing, manufacturing, uses and labelling of fats in the food supply. *Annals of Nutrition & Metabolism* 55 (1–3):273–300. https://www.jstor.org/stable/48514102. doi: 10.1159/000229006.

Hernández-Martín, E., and C. Otero. 2008. Different enzyme requirements for the synthesis of biodiesel: Novozym 435 and Lipozyme TL IM. *Bioresource Technology* 99 (2):277–86. doi: 10.1016/j.biortech.2006.12.024.

Hiol, A., Marie, D. Jonzo, N. Rugani, D. Druet, L. Sarda, and L. C. Comeau. 2000. Purification and characterization of an extracellular lipase from a thermophilic *Rhizopus oryzae* strain isolated from palm fruit. *Enzyme and Microbial Technology* 26 (5–6):421–30. doi.org/ (99)00:421–30. 8 doi: 10.1016/S0141-0229.

Horchani, H., I. Aissa, S. Ouertani, Z. Zarai, Y. Gargouri, and A. Sayari. 2012. Staphylococcal lipases: Biotechnological applications. *Journal of Molecular Catalysis B: Enzymatic* 76:125–32. doi:10.1016/j.molcatb.2011.11.018.

Horchani, H., N. B. Salem, Z. Zarai, A. Sayari, Y. Gargouri, and M. Chaâbouni. 2010. Enzymatic synthesis of eugenol benzoate by immobilized *Staphylococcus aureus* lipase: Optimization using response surface methodology and determination of antioxidant activity. *Bioresource Technology* 101 (8):2809–17. doi: 10.1016/j.biortech.2009.10.082.

Hotta, Y., S. Ezaki, H. Atomi, and T. Imanaka. 2002. Extremely stable and versatile carboxylesterase from a hyperthermophilic archaeon. *Applied and Environmental Microbiology* 68 (8):3925–31. doi: 10.1128/AEM.68.8.3925-3931.2002.

Houde, A., A. Kademi, and D. Leblanc. 2004. Lipases and their industrial applications: an overview. *Applied Biochemistry and Biotechnology* 118 (1–3):155–70. doi: 10.1385/ABAB:118:1-3:155.

Hu, J., W. Cai, C. Wang, X. Du, J. Lin, and J. Cai. 2018. Purification and characterization of alkaline lipase production by *Pseudomonas aeruginosa* HFE733 and application for biodegradation in food wastewater treatment. *Biotechnology & Biotechnological Equipment* 32 (3):583–90. doi: 10.1080/13102818.2018.1446764.

Huang, Z., L. Stipkovits, H. Zheng, L. Serventi, and C. S. Brennan. 2019. Bovine milk fats and their replacers in baked goods: A review. *Foods* 8 (9):383. doi: 10.3390/foods8090383.

Husain, Q., S. A. Ansari, F. Alam, and A. Azam. 2011. Immobilization of Aspergillus oryzae β galactosidase on zinc oxide nanoparticles via simple adsorption mechanism. *International Journal of Biological Macromolecules* 49 (1):37–43. doi: 10.1016/j.ijbiomac.2011.03.011.

Hwang, S., J. Ahn, S. Lee, T. G. Lee, S. Haam, K. Lee, I.-S. Ahn, and J. Jung. 2004. Evaluation of cellulose-binding domain fused to a lipase for the lipase immobilization. *Biotechnology Letters* 26 (7):603–5. doi: 10.1023/B:BILE.0000021964.69500.6f.

Hwang, H. T., F. Qi, C. Yuan, X. Zhao, D. Ramkrishna, D. Liu, and A. Varma. 2014. Lipase-catalyzed process for biodiesel production: protein engineering and lipase production . *Biotechnology and Bioengineering* 111 (4):639–53. doi: 10.1002/bit.25162.

Ianni, A., F. Bennato, C. Martino, L. Grotta, and G. Martino. 2020. Volatile flavor compounds in cheese as affected by ruminant diet. *Molecules* 25 (3):461. doi: 10.3390/molecules25030461.

Ingenbosch, K. N., A. Rousek, D. S. Wunschik, and K. Hoffmann-Jacobsen. 2019. A fluorescence-based activity assay for immobilized lipases in non-native media. *Analytical Biochemistry* 569:22–7. doi: 10.1016/j.ab.2019.01.005.

Ishak, S. N. H., M. Masomian, N. H. A. Kamarudin, M. S. M. Ali, T. C. Leow, and R. N. Z. R. Abd Rahman. 2019. Changes of thermostability, organic solvent, and pH stability in *Geobacillus zalihae* HT1 and its mutant by calcium ion. *International Journal of Molecular Sciences* 20 (10):2561. doi: 10.3390/ijms20102561.

Ishchenko, A., L. Peng, E. Zinovev, A. Vlasov, S. C. Lee, A. Kuklin, A. Mishin, V. Borshchevskiy, Q. Zhang, and V. Cherezov. 2017. Chemically stable lipids for membrane protein crystallization. *Crystal Growth & Design* 17 (6):3502–11. doi: 10.1021/acs.cgd.7b00458.

Ismail, A. R., and K.-H. Baek. 2020. Lipase immobilization with support materials, preparation techniques, and applications: Present

and future aspects. *International Journal of Biological Macromolecules* 163:1624–39. doi: 10.1016/j.ijbiomac.2020.09.021.

Isobe, K., K. Nokihara, S. Yamaguchi, T. Mase, and R. D. Schmid. 1992. Crystallization and characterization of monoacylglycerol and diacylglycerol lipase from *Penicillium camembertii*. *European Journal of Biochemistry* 203 (1–2):233–7. doi: 10.1111/j.1432-1033.1992.tb19851.x.

Ittrat, P., T. Chacho, J. Pholprayoon, N. Suttiwarayanon, and J. Charoenpanich. 2014. Application of agriculture waste as a support for lipase immobilization. *Biocatalysis and Agricultural Biotechnology* 3 (3):77–82. doi: 10.1016/j.bcab.2014.02.002.

Iyer, P. V., and L. Ananthanarayan. 2008. Enzyme stability and stabilization—aqueous and non-aqueous environment. *Process Biochemistry* 43 (10):1019–32. doi: 10.1016/j.procbio.2008.06.004.

Jaeger, K.-E., and M. T. Reetz. 1998. Microbial lipases form versatile tools for biotechnology. *Trends in Biotechnology* 16 (9):396–403. doi: 10.1016/S0167-7799. (98)01195-0

Jaenicke, R., and G. Böhm. 1998. The stability of proteins in extreme environments. *Current Opinion in Structural Biology* 8 (6):738–48. doi.org/10.1016/S0959-440X (98)80094-8 doi: 10.1016/S0959-440X(98)80094-8.

Jancsik, V., Z. Beleznai, and T. Keleti. 1982. Enzyme immobilization by poly (vinyl alcohol) gel entrapment. *Journal of Molecular Catalysis* 14 (3):297–306. doi.org/ (82)80090-4 doi: 10.1016/0304-5102.

Javed, S., F. Azeem, S. Hussain, I. Rasul, M. H. Siddique, M. Riaz, M. Afzal, A. Kouser, and H. Nadeem. 2018. Bacterial lipases: A review on purification and characterization. *Progress in Biophysics and Molecular Biology* 132:23–34. doi: 10.1016/j.pbiomolbio.2017.07.014.

Jayathilakan, K., K. Sultana, K. Radhakrishna, and A. S. Bawa. 2012. Utilization of byproducts and waste materials from meat, poultry and fish processing industries: a review. *Journal of Food Science and Technology* 49 (3):278–93. doi: 10.1007/s13197-011-0290-7.

Jiao, L., Q. Zhou, Z. Su, L. Xu, and Y. Yan. 2018. High-level extracellular production of *Rhizopus oryzae* lipase in *Pichia pastoris* via a strategy combining optimization of gene-copy number with co-expression of ERAD-related proteins. *Protein Expression and Purification* 147:1–12. doi: 10.1016/j.pep.2018.02.005.

Johnson, P. A., H. J. Park, and A. J. Driscoll. 2011. Enzyme nanoparticle fabrication: Magnetic nanoparticle synthesis and enzyme immobilization. In *Enzyme stabilization and immobilization. Methods in molecular biology (methods and protocols)*, S. Minteer, vol 679. Totowa, NJ: Humana Press. doi: 10.1007/978-1-60761-895-9_15.

Jun, L. Y., L. S. Yon, N. M. Mubarak, C. H. Bing, S. Pan, M. K. Danquah, E. C. Abdullah, and M. Khalid. 2019. An overview of immobilized enzyme technologies for dye and phenolic removal from wastewater. *Journal of Environmental Chemical Engineering* 7 (2):102961. doi: 10.1016/j.jece.2019.102961.

Kahveci, D., and X. Xu. 2011. Enhancement of activity and selectivity of *Candida rugosa* lipase and *Candida antarctica* lipase A by bioimprinting and/or immobilization for application in the selective ethanolysis of fish oil. *Biotechnology Letters* 33 (10):2065–71. doi: 10.1007/s10529-011-0671-z.

Kambourova, M., N. Kirilova, R. Mandeva, and A. Derekova. 2003. Purification and properties of thermostable lipase from a thermophilic *Bacillus stearothermophilus* MC 7. *Journal of Molecular Catalysis B: Enzymatic* 22 (5–6):307–13. doi: 10.1016/S1381-1177(03)00045-6.

Kang, J.-H., and F. Kondo. 2003. Determination of bisphenol A in milk and dairy products by high-performance liquid chromatography with fluorescence detection. *Journal of Food Protection* 66 (8):1439–43. doi: 10.4315/0362-028x-66.8.1439.

Kanimozhi, S., and K. Perinbam. 2015. Optimization of media components and growth conditions to enhance lipase production by *Pseudomonas* sp. Lp1. *Biomedical and Pharmacology Journal* 3 (2):329–38. http://biomedpharmajournal.org/?p=1562.

Kanmani, P., J. Aravind, and K. Kumaresan. 2015. An insight into microbial lipases and their environmental facet. *International Journal of Environmental Science and Technology* 12 (3):1147–62. doi: 10.1007/s13762-014-0605-0.

Kanmani, P., K. Kumaresan, and J. Aravind. 2015. Gene cloning, expression, and characterization of the *Bacillus amyloliquefaciens* PS35 lipase. *Brazilian Journal of Microbiology: [Publication of the Brazilian Society for Microbiology]* 46 (4):1235–43. doi: 10.1590/S1517-838246420141068.

Karbalaei, M., S. A. Rezaee, and H. Farsiani. 2020. *Pichia pastoris*: A highly successful expression system for optimal synthesis of heterologous proteins. *Journal of Cellular Physiology* 235 (9):5867–81. doi: 10.1002/jcp.29583.

Kartal, F., Ç. Ali Kilin, and S. Timur. 2007. Lipase biosensor for tributyrin and pesticide detection. *International Journal of Environmental Analytical Chemistry* 87 (10–11):715–22. doi: 10.1080/03067310701327741.

Katchalski-Katzir, E., and D. M. Kraemer. 2000. Eupergit® C, a carrier for immobilization of enzymes of industrial potential. *Journal of Molecular Catalysis B: Enzymatic* 10 (1–3):157–76. doi: 10.1016/S1381-1177(00)00124-7.

Kato, M., J. Fuchimoto, T. Tanino, A. Kondo, H. Fukuda, and M. Ueda. 2007. Preparation of a whole-cell biocatalyst of mutated *Candida antarctica* lipase B (mCALB) by a yeast molecular display system and its practical properties. *Applied Microbiology and Biotechnology* 75 (3):549–55. doi: 10.1007/s00253-006-0835-2.

Kauffmann, I., and C. Schmidt-Dannert. 2001. Conversion of *Bacillus thermocatenulatus* lipase into an efficient phospholipase with increased activity towards long-chain fatty acyl substrates by directed evolution and rational design. *Protein Engineering* 14 (11):919–28. doi: 10.1093/protein/14.11.919.

Khan, M. T., A. C. Kaushik, Q. u a. Rana, S. I. Malik, A. S. Khan, D.-Q. Wei, W. S. S. Ahmad, S. Ali, and M. Irfan. 2020. Characterization and synthetic biology of lipase from *Bacillus amyloliquefaciens* strain. *Archives of Microbiology* 202 (6):1497–506. doi: 10.1007/s00203-020-01869-0.

Kilara, A., Khem, M. Shahani, Triveni, and P. Shukla. 1979. The use of immobilized enzymes in the food industry: A review. *CRC Critical Reviews in Food Science and Nutrition* 12 (2):161–98. doi: 10.1080/10408397909527276.

Kilcawley, K. N., Martin, G. Wilkinson, and P. F. Fox. 1998. Enzyme-modified cheese. *International Dairy Journal* 8 (1):1–10. doi: 10.1016/S0958-6946(98)00010-7.

Kilcawley, K. N., M. G. Wilkinson, and P. F. Fox. 2001. A survey of lipolytic and glycolytic end-products in commercial Cheddar enzyme-modified cheese. *Journal of Dairy Science* 84 (1):66–73. doi: 10.3168/jds.S0022-0302. (01)74453-0

Kim, D., and A. E. Herr. 2013. Protein immobilization techniques for microfluidic assays. *Biomicrofluidics* 7 (4):41501. doi: 10.1063/1.4816934.

Kimiyasu, I., T. Akiba, and S. Yamaguchi. 1988. Crystallization and characterization of lipase from *Penicillium cyclopium*. *Agricultural and Biological Chemistry* 52 (1):41–7. doi.org/10.1080/00021369.1988.10868607 doi: 10.1271/bbb1961.52.41.

Kivanc, M., M. Yilmaz, and F. Demir. 2011. The occurrence of *Aeromonas* in drinking water, tap water and the porsuk river. *Brazilian Journal of Microbiology: [Publication of the Brazilian Society for Microbiology]* 42 (1):126–31. doi: 10.1590/S1517-83822011000100016.

Klein, R. R., G. King, R. A. Moreau, and M. J. Haas. 1997. Altered acyl chain length specificity of *Rhizopus delemar* lipase through mutagenesis and molecular modeling. *Lipids* 32 (2):123–30. doi: 10.1007/s11745-997-0016-1.

Klibanov, A. M. 1983. Immobilized enzymes and cells as practical catalysts. *Science (New York, NY)* 219 (4585):722–7. doi: 10.1126/science.219.4585.722.

Klonoff, D. C. 2007. Replacements for trans fats-will there be an oil shortage? *Journal of Diabetes Science and Technology* 1 (3):415–22. doi: 10.1177/193229680700100316.

Kluger, R., and A. Alagic. 2004. Chemical cross-linking and protein-protein interactions-a review with illustrative protocols. *Bioorganic Chemistry* 32 (6):451–72. doi: 10.1016/j.bioorg.2004.08.002.

Kobayashi, T. 2011. Lipase-catalyzed syntheses of sugar esters in non-aqueous media. *Biotechnology Letters* 33 (10):1911–9. doi: 10.1007/s10529-011-0663-z.

Kojima, Y., and S. Shimizu. 2003. Purification and characterization of the lipase from *Pseudomonas fluorescens* HU380. *Journal of Bioscience and Bioengineering* 96 (3): 219–226. doi: 10.1016/S1389-1723(03)80185-8.

Kovacic, F., B. Nikolina, K. Ulrich, and K. Jaeger. 2019. Classification of lipolytic enzymes from bacteria. Aerobic utilization of hydrocarbons, oils, and lipids. In *Handbook of hydrocarbon and lipid microbiology*, vol. 24, 255–89. Cham: Springer. doi: 10.1007/978-3-319-50418-6.

Krajewska, B. 2004. Application of chitin-and chitosan-based materials for enzyme immobilizations: A review. *Enzyme and Microbial Technology* 35 (2–3):126–39. doi: 10.1016/j.enzmictec.2003.12.013.

Krebs, J. E., P. Vaishampayan, A. J. Probst, L. M. Tom, V. T. Marteinsson, G. L. Andersen, and K. Venkateswaran. 2014. Microbial community structures of novel Icelandic hot spring systems revealed by PhyloChip G3 analysis. *Astrobiology* 14 (3):229–40. doi: 10.1089/ast.2013.1008.

Krieger, N., M. Angela Taipa, M. Raquel Aires-Barros, E. H. M. Melo, J. L. Lima-Filho, and J. M. S. Cabral. 1997. Purification of the *Penicillium citrinum* lipase using AOT reversed micelles. *Journal of Chemical Technology & Biotechnology* 69 (1):77–85. doi: 10.1002/(SICI)1097-4660(199705)69:1<77::AID-JCTB666>3.0.CO;2-V.

Krystallis, C., G. S. Masterton, P. C. Hayes, and J. N. Plevris. 2012. Update of endoscopy in liver disease: More than just treating varices. *World Journal of Gastroenterology* 18 (5):401–11. doi: 10.3748/wjg.v18.i5.401.

Krzeslak, J., G. Gerritse, R. V. Merkerk, R. H. Cool, and W. J. Quax. 2008. Lipase expression in *Pseudomonas alcaligenes* is under the control of a two-component regulatory system. *Applied and Environmental Microbiology* 74 (5):1402–11. doi: 10.1128/AEM.01632-07.

Kujawa, J., M. Głodek, I. Koter, B. Ośmiałowski, K. Knozowska, S. Al-Gharabli, L. F. Dumée, and W. Kujawski. 2021. Molecular decoration of ceramic supports for highly effective enzyme immobilization—Material approach. *Materials* 14 (1):201. doi: 10.3390/ma14010201.

Kuksis, A., and J. M. R. Beveridge. 1960. Preparation and certain physical properties of some plant steryl esters. *The Journal of Organic Chemistry* 25 (7):1209–19. doi: 10.1021/jo01077a035.

Kundys, A., E. B. Florjańczyk, A. Fabiszewska, and J. Małajowicz. 2018. *Candida antarctica* lipase B as a catalyst for cyclic esters synthesis, their polymerization, and degradation of aliphatic polyesters. *Journal of Polymers and the Environment* 26 (1):396–407. doi: 10.1007/s10924-017-0945-1.

Kurtovic, I., S. N. Marshall, X. Zhao, and B. K. Simpson. 2009. Lipases from mammals and fishes. *Reviews in Fisheries Science* 17 (1):18–40. doi: 10.1080/10641260802031322.

Lan, D., M. Qu, B. Yang, and Y. Wang. 2016. Enhancing production of lipase MAS1 from marine *Streptomyces* sp. strain in *Pichia pastoris* by chaperones co-expression. *Electronic Journal of Biotechnology* 22 (2016):62–7. doi: 10.1016/j.ejbt.2016.06.003.

Larios, A., H. S. García, R. M. Oliart, and G. Valerio-Alfaro. 2004. Synthesis of flavor and fragrance esters using *Candida antarctica* lipase. *Applied Microbiology and Biotechnology* 65 (4):373–6. doi: 10.1007/s00253-004-1602-x.

Lee, B.-M., J.-H. Choi, S. I. Hong, S. W. Yoon, B. H. Kim, C.-T. Kim, C.-J. Kim, Y. Kim, and I.-H. Kim. 2011. Enrichment of pinolenic acid from pine nut oil via lipase-catalyzed ethanolysis with an immobilized *Candida antarctica* lipase. *Biocatalysis and Biotransformation* 29 (4):155–60. doi: 10.3109/10242422.2011.590983.

Lee, B. H., K. N. Kilcawley, J. A. Hannon, S. Y. Park, M. G. Wilkinson, and T. P. Beresford. 2007. The use of viable and heat-shocked *Lactobacillus helveticus* DPC 4571 in enzyme-modified cheese production. *Food Biotechnology* 21 (2):129–43. doi: 10.1080/08905430701410530.

Lee, D.-W., H.-W. Kim, K.-W. Lee, B.-C. Kim, E.-A. Choe, H.-S. Lee, D.-S. Kim, and Y.-R. Pyun. 2001. Purification and characterization of two distinct thermostable lipases from the gram-positive thermophilic bacterium *Bacillus thermoleovorans* ID-1. *Enzyme and Microbial Technology* 29 (6–7):363–71. doi.org/ (01)00:363–71. 2 doi: 10.1016/S0141-0229.

Lee, C.-H., T. Lin, and C.-Y. Mou. 2009. Mesoporous materials for encapsulating enzymes. *Nano Today* 4 (2):165–79. doi: 10.1016/j.nantod.2009.02.001.

Lee, E. Y., and M. L. Shuler. 2007. Molecular engineering of epoxide hydrolase and its application to asymmetric and enantioconvergent hydrolysis. *Biotechnology and Bioengineering* 98 (2):318–27. doi: 10.1002/bit.21444.

Lemieux, L., and R. E. Simard. 1991. Bitter flavor in dairy products. I. A review of the factors likely to influence its development, mainly in cheese manufacture. *Le Lait* 71 (6):599–636. doi: 10.1051/lait:1991647.

Lennen, R. M., and B. F. Pfleger. 2012. Engineering *Escherichia coli* to synthesize free fatty acids. *Trends in Biotechnology* 30 (12):659–67. doi: 10.1016/j.tibtech.2012.09.006.

Levisson, M., J. v d. Oost, and S. W. M. Kengen. 2009. Carboxylic ester hydrolases from hyperthermophiles. *Extremophiles: Life under Extreme Conditions* 13 (4):567–81. doi: 10.1007/s00792-009-0260-4.

Lewis, J., Y. C. Lin, L. K. Royston, and R. C. Thompson. 1965. 1194. The chemistry of polynuclear compounds. Part III. Magnetic properties of some carboxylic acid derivatives of copper(II). *Journal of the Chemical Society (Resumed)* 1965:6464. doi:10.1039/jr9650006464.

Liang, S., X.-L. Wu, J. Xiong, M.-H. Zong, and W.-Y. Lou. 2020. Metal-organic frameworks as novel matrices for efficient enzyme immobilization: An updated review. *Coordination Chemistry Reviews* 406 (213149):213149. doi: 10.1016/j.ccr.2019.213149.

Liebeton, K., A. Zacharias, and K. E. Jaeger. 2001. Disulfide bond in *Pseudomonas aeruginosa* lipase stabilizes the structure but is not required for interaction with its foldase. *Journal of Bacteriology* 183 (2):597–603. doi: 10.1128/JB.183.2.597-603.2001.

Li, Z., W. Leung, A. Yon, J. Nguyen, V. C. Perez, J. Vu, W. Giang, L. T. Luong, T. Phan, K. A. Salazar, et al. 2010. Secretion and proteolysis of heterologous proteins fused to the *Escherichia coli* maltose binding protein in Pichia pastoris . *Protein Expression and Purification* 72 (1):113–24. doi: 10.1016/j.pep.2010.03.004.

Li, H.-C., and S.-Y. Lo. 2015. Hepatitis C virus: Virology, diagnosis and treatment. *World Journal of Hepatology* 7 (10):1377–89. doi: 10.4254/wjh.v7.i10.1377.

Lima, G. V., M. R. da Silva, T. de Sousa Fonseca, L. B. de Lima, M. d C. F. de Oliveira, T. L. G. de Lemos, D. Zampieri, J. C. S. dos Santos, N. S. Rios, L. R. B. Gonçalves, et al. 2017. Chemoenzymatic synthesis of (S)-Pindolol using lipases. *Applied Catalysis A: General* 546:7–14. doi: 10.1016/j.apcata.2017.08.003.

Lim, D. S., S. J. Kwack, K.-B. Kim, H. S. Kim, and B. M. Lee. 2009. Potential risk of bisphenol A migration from polycarbonate containers after heating, boiling, and microwaving. *Journal of Toxicology and Environmental Health. Part A* 72 (21–22):1285–91. doi: 10.1080/15287390903212329.

Lim, H. J., and J. H. Lee. 2012. New diagnostic methods for tuberculosis. *Korean Journal of Medicine* 82 (3):263–8. doi: 10.1109/5.771073.

Li, Y., and D. J. McClements. 2011. Inhibition of lipase-catalyzed hydrolysis of emulsified triglyceride oils by low-molecular weight surfactants under simulated gastrointestinal conditions. *European Journal of Pharmaceutics and Biopharmaceutics: Official Journal of Arbeitsgemeinschaft Fur Pharmazeutische Verfahrenstechnik e.V* 79 (2):423–31. doi: 10.1016/j.ejpb.2011.03.019.

Lin, X.-S., K.-H. Zhao, Q.-L. Zhou, K.-Q. Xie, P. J. Halling, and Z. Yang. 2016. *Aspergillus oryzae* lipase-catalyzed synthesis of glucose laurate with excellent productivity. *Bioresources and Bioprocessing* 3 (1):1–7. doi: 10.1186/s40643-015-0080-6.

Liu, X., X. Chen, Y. Li, X. Wang, X. Peng, and W. Zhu. 2012. Preparation of superparamagnetic Fe3O4@alginate/chitosan nanospheres for Candida rugosa lipase immobilization and utilization of layer-by-layer assembly to enhance the stability of immobilized lipase. *ACS Applied Materials & Interfaces* 4 (10):5169–78. doi: 10.1021/am301104c.

Liu, W., B. Jia, H. Zhao, L. Xu, and Y. Yan. 2010. Preparation of a whole-cell biocatalyst of *Aspergillus niger* lipase and its practical

properties. *Journal of Agricultural and Food Chemistry* 58 (19):10426–30. doi: 10.1021/jf1008555.

Liu, D., R. D. Schmid, and M. Rusnak. 2006. Functional expression of *Candida antarctica* lipase B in the *Escherichia coli* cytoplasm-a screening system for a frequently used biocatalyst. *Applied Microbiology and Biotechnology* 72 (5):1024–32. doi: 10.1007/s00253-006-0369-7.

Liu, L., H. Yang, H.-d. Shin, R. R. Chen, J. Li, G. Du, and J. Chen. 2013. How to achieve high-level expression of microbial enzymes: Strategies and perspectives. *Bioengineered* 4 (4):212–23. doi: 10.4161/bioe.24761.

Li, W., and J. Zhang. 2016. Recent developments in the synthesis and utilization of chiral β-aminophosphine derivatives as catalysts or ligands. *Chemical Society Reviews* 45 (6):1657–77. doi: 10.1039/c5cs00469a.

Li, P.-Y., Y.-Q. Zhang, Y. Zhang, W.-X. Jiang, Y.-J. Wang, Y.-S. Zhang, Z.-Z. Sun, C.-Y. Li, Y.-Z. Zhang, M. Shi, et al. 2020. Study on a novel cold-active and halotolerant monoacylglycerol lipase widespread in marine bacteria reveals a new group of bacterial monoacylglycerol lipases containing unusual C(A/S)HSMG catalytic motifs. *Frontiers in Microbiology* 11:9. doi: 10.3389/fmicb.2020.00009.

Li, N., and M.-H. Zong. 2010. Lipases from the genus *Penicillium*: Production, purification, characterization, and applications. *Journal of Molecular Catalysis B: Enzymatic* 66 (1–2):43–54. doi: 10.1016/j.molcatb.2010.05.004.

Löbs, A.-K., C. Schwartz, and I. Wheeldon. 2017. Genome and metabolic engineering in non-conventional yeasts: Current advances and applications. *Synthetic and Systems Biotechnology* 2 (3):198–207. doi: 10.1016/j.synbio.2017.08.002.

Lobstein, J., C. A. Emrich, C. Jeans, M. Faulkner, P. Riggs, and M. Berkmen. 2012. SHuffle, a novel Escherichia coli protein expression strain capable of correctly folding disulfide bonded proteins in its cytoplasm. *Microbial Cell Factories* 11 (1):56–16. doi: 10.1186/1475-2859-11-56.

Longo, M. A., and D. Combes. 1997. Influence of surface hydrophilic/hydrophobic balance on enzyme properties. *Journal of Biotechnology* 58 (1):21–32. doi: 10.1016/s0168-1656(97)00120-x.

Lopes, D. B., L. P. Fraga, L. F. Fleuri, and G. A. Macedo. 2011. Lipase and esterase: To what extent can this classification be applied accurately?. *Ciência e Tecnologia de Alimentos* 31 (3):603–13. doi: 10.1590/S0101-20612011000300009.

López-Serrano, P., L. Cao, F. V. Rantwijk, and R. A. Sheldon. 2002. Cross-linked enzyme aggregates with enhanced activity: Application to lipases. *Biotechnology Letters* 24 (16):1379–83. doi: 10.1023/A:1019863314646.

Lu, J., C. J. Brigham, C. Rha, and A. J. Sinskey. 2013. Characterization of an extracellular lipase and its chaperone from *Ralstonia eutropha* H16. *Applied Microbiology and Biotechnology* 97 (6):2443–54. doi: 10.1007/s00253-012-4115-z.

Lund, T., and P. E. Granum. 1996. Characterisation of a non-haemolytic enterotoxin complex from Bacillus cereus isolated after a foodborne outbreak . *FEMS Microbiology Letters* 141 (2–3):151–6. doi: 10.1111/j.1574-6968.1996.tb08377.x.

Ma, B., L. Cheong, X. Weng, C.-P. Tan, and C. Shen. 2018. Lipase@ZIF-8 nanoparticles-based biosensor for direct and sensitive detection of methyl parathion. *Electrochimica Acta* 283:509–16. doi: 10.1016/j.electacta.2018.06.176.

Machado, S. G., F. Baglinière, S. Marchand, E. V. Coillie, M. C. D. Vanetti, J. D. Block, and M. Heyndrickx. 2017. The biodiversity of the microbiota producing heat-resistant enzymes responsible for spoilage in processed bovine milk and dairy products. *Frontiers in Microbiology* 8:302. doi: 10.3389/fmicb.2017.00302.

Macrae, A. R. 1983. Lipase-catalyzed interesterification of oils and fats. *Journal of the American Oil Chemists' Society* 60 (2Part1):291–4. doi: 10.1007/BF02543502.

Macrae, A. R., and R. C. Hammond. 1985. Present and future applications of lipases. *Biotechnology and Genetic Engineering Reviews* 3 (1):193–218. doi: 10.1080/02648725.1985.10647813.

Madzak, C., C. Gaillardin, and J. Beckerich. 2004. Heterologous protein expression and secretion in the non-conventional yeast *Yarrowia lipolytica*: A review. *Journal of Biotechnology* 109 (1–2):63–81. doi: 10.1016/j.jbiotec.2003.10.027.

Mallardi, A., V. Angarano, M. Magliulo, L. Torsi, and G. Palazzo. 2015. General approach to the immobilization of glycoenzyme chains inside calcium alginate beads for bioassay. *Analytical Chemistry* 87 (22):11337–44. doi: 10.1021/acs.analchem.5b02636.

Maruyama, T., M. Nakajima, S. Uchikawa, H. Nabetani, S. Furusaki, and M. Seki. 2000. Oil-water interfacial activation of lipase for interesterification of triglyceride and fatty acid. *Journal of the American Oil Chemists' Society* 77 (11):1121. doi: 10.1007/s11746-000-0176-4.

Mateo, C., J. M. Palomo, G. Fernandez-Lorente, J. M. Guisan, and R. Fernandez-Lafuente. 2007. Improvement of enzyme activity, stability, and selectivity via immobilization techniques. *Enzyme and Microbial Technology* 40 (6):1451–63. doi: 10.1016/j.enzmictec.2007.01.018.

Matsumoto, M., and K. Ohashi. 2003. Effect of immobilization on the thermostability of lipase from *Candida rugosa*. *Biochemical Engineering Journal* 14 (1):75–7. doi.org/ (02)00:75–7. 9 doi: 10.1016/S1369-703X.

Matull, W. R., S. P. Pereira, and J. W. O'Donohue. 2006. Biochemical markers of acute pancreatitis. *Journal of Clinical Pathology* 59 (4):340–4. doi: 10.1136/jcp.2002.002923.

Mayer, F. L., D. Wilson, and B. Hube. 2013. *Candida albicans* pathogenicity mechanisms. *Virulence* 4 (2):119–28. doi: 10.4161/viru.22913.

McCutcheon, A. D. 2000. Neurological damage and duodenopancreatic reflux in the pathogenesis of alcoholic pancreatitis. *Archives of Surgery (Chicago, IL: 1960)* 135 (3):278–85. doi: 10.1001/archsurg.135.3.278.

McMahon, D. J., R. J. Brown, and C. A. Ernstrom. 1984. Enzymic coagulation of casein micelles: A review. *Journal of Dairy Science* 67 (4):745–8. doi: 10.3168/jds.S0022-0302(84)81390-9.

McMenamin, J. D., T. M. Zaccone, T. Coenye, P. Vandamme, and J. J. LiPuma. 2000. Misidentification of *Burkholderia cepacia* in US cystic fibrosis treatment centers: an analysis of 1,051 recent sputum isolates. *Chest* 117 (6):1661–5. doi: 10.1378/chest.117.6.1661.

McSweeney, P. L. H. 2004. Biochemistry of cheese ripening. *International Journal of Dairy Technology* 57 (2–3):127–44. doi: 10.1111/j.1471-0307.2004.00147.x.

Mead, J. R., S. A. Irvine, and D. P. Ramji. 2002. Lipoprotein lipase: Structure, function, regulation, and role in disease. *Journal of Molecular Medicine (Berlin, Germany)* 80 (12):753–69. doi: 10.1007/s00109-002-0384-9.

Meadows, C. W., A. Kang, and T. S. Lee. 2018. Metabolic engineering for advanced biofuels production and recent advances toward commercialization. *Biotechnology Journal* 13 (1):1600433. doi: 10.1002/biot.201600433.

Mehta, A., U. Bodh, and R. Gupta. 2018. Isolation of a novel lipase producing fungal isolate *Aspergillus fumigatus* and production optimization of the enzyme. *Biocatalysis and Biotransformation* 36 (6):450–7. doi: 10.1080/10242422.2018.1447565.

Meunchan, M., S. Michely, H. Devillers, J.-M. Nicaud, A. Marty, and C. Neuvéglise. 2015. Comprehensive analysis of a yeast lipase family in the *Yarrowia clade*. *PloS One* 10 (11):e0143096. doi: 10.1371/journal.pone.0143096.

Meunier, S. M., and R. L. Legge. 2013. Study of support materials for sol-gel immobilized lipase. *Biocatalysis and Biotransformation* 31 (4):190–6. doi: 10.3109/10242422.2013.815744.

Mine, Y. 1998. Adsorption behavior of egg yolk low-density lipoproteins in oil-in-water emulsions. *Journal of Agricultural and Food Chemistry* 46 (1):36–41. doi: 10.1021/jf970306y.

Miranda, J. M., X. Anton, C. Redondo-Valbuena, P. Roca-Saavedra, J. A. Rodriguez, A. Lamas, C. M. Franco, and A. Cepeda. 2015. Egg and egg-derived foods: Effects on human health and use as functional foods. *Nutrients* 7 (1):706–29. doi: 10.3390/nu7010706.

Misson, M., H. Zhang, and B. Jin. 2015. Nanobiocatalyst advancements and bioprocessing applications. *Journal of the Royal Society, Interface* 12 (102):20140891. doi: 10.1098/rsif.2014.0891.

Moatsou, G., E. Zoidou, E. Choundala, K. Koutsaris, O. Kopsia, K. Thergiaki, and L. Sakkas. 2019. Development of reduced-fat,

reduced-sodium semi-hard sheep milk cheese. *Foods* 8 (6):204. doi: 10.3390/foods8060204.

Mobarak-Qamsari, E., R. Kasra-Kermanshahi, and Z. Moosavi-Nejad. 2011. Isolation and identification of a novel, lipase-producing bacterium, *Pseudomonas aeruginosa* KM110. *Iranian Journal of Microbiology* 3 (2):92–8. PMID: 22347589.

Mohamed, S. S., H. M. Ahmed, M. A. El-Bendary, M. E. Moharam, and H. A. Amin. 2021. Response surface methodology for optimization of *Rhizopus stolonifer* 1aNRC11 mutant F whole-cell lipase production as a biocatalyst for methanolysis of waste frying oil. *Biocatalysis and Biotransformation* 39 (3):232–40. doi: 10.1080/10242422.2020.1869218.

Mohebbi, M., J. Barouei, M. R. Akbarzadeh-T, A. R. Rowhanimanesh, M. B. Habibi-Najafi, and M. Yavarmanesh. 2008. Modeling and optimization of viscosity in enzyme-modified cheese by fuzzy logic and genetic algorithm. *Computers and Electronics in Agriculture* 62 (2):260–5. doi: 10.1016/j.compag.2008.01.010.

Mokhtar, N. F., R. N. Z. R. Abd. Rahman, N. D. Muhd Noor, F. Mohd Shariff, and M. S. Mohamad Ali. 2020. The immobilization of lipases on porous support by adsorption and hydrophobic interaction method. *Catalysts* 10 (7):744. doi: 10.3390/catal10070744.

Moradali, M. F., S. Ghods, and B. H. A. Rehm. 2017. *Pseudomonas aeruginosa* lifestyle: A paradigm for adaptation, survival, and persistence. *Frontiers in Cellular and Infection Microbiology* 7:39. doi: 10.3389/fcimb.2017.00039.

Moreno-Pérez, S., J. M. Guisan, and G. Fernandez-Lorente. 2014. Selective ethanolysis of fish oil catalyzed by immobilized lipases. *Journal of the American Oil Chemists' Society* 91 (1):63–9. doi: 10.1007/s11746-013-2348-3.

Muhamad, O. A.-L., K. M. Khleifat, K. Y. Alsharafa, H. N. Qaralleh, and S. A. Alrawashdeh. 2019. Purification and characterization of a mesophilic organic solvent tolerant lipase produced by *Acinetobacter* sp. K5b4. *Biocatalysis and Biotransformation* 37 (2):139–51. doi: 10.1080/10242422.2018.1506445.

Muniandy, M., O. Lasekan, H. M. Ghazali, and M. Rahman. 2019. Lipase-catalyzed formation of pentyl nonanoate using screened immobilized lipase from *Rhizomucor meihei*. *Brazilian Journal of Chemical Engineering* 36 (3):1089–97. doi: 10.1590/0104-6632.20190363s20180419.

Muralidhar, R. V., R. R. Chirumamilla, R. Marchant, V. N. Ramachandran, O. P. Ward, and P. Nigam. 2002. Understanding lipase stereoselectivity. *World Journal of Microbiology and Biotechnology* 18 (2):81–97. doi: 10.1023/A:1014417223956.

Murty, V. R., J. Bhat, and P. K. A. Muniswaran. 2002. Hydrolysis of oils by using immobilized lipase enzyme: A review. *Biotechnology and Bioprocess Engineering* 7 (2):57–66. doi: 10.1007/BF02935881.

Nagaoka, K., and Y. Yamada. 1973. Purification of Mucor lipases and their properties. *Agricultural and Biological Chemistry* 37 (12):2791–6. doi.org/10.1271/bbb1961.37.2791 doi: 10.1080/00021369.1973.10861078.

Nagaroor, V., and S. N. Gummadi. 2020. Biochemical characterization of an esterase from *Clostridium acetobutylicum* with novel GYSMG pentapeptide motif at the catalytic domain. *Journal of Industrial Microbiology & Biotechnology* 47 (2):169–81. doi: 10.1007/s10295-019-02253-8.

Naglik, J. R., J. P. Richardson, and D. L. Moyes. 2014. *Candida albicans* pathogenicity and epithelial immunity. *PLoS Pathogens* 10 (8):e1004257. doi: 10.1371/journal.ppat.1004257.

Nakai, S., W. A. Blair, A. Helmersen, and B. A. Eagles. 1968. Desalting milk and whey by ion retardation and gel filtration. *Journal of Dairy Science* 51 (12):1909–11. doi.org/ (68)87310-2 doi: 10.3168/jds.S0022-0302.

Narayanan, N., M. Khan, and C. P. Chou. 2010. Enhancing functional expression of heterologous lipase B in *Escherichia coli* by extracellular secretion. *Journal of Industrial Microbiology & Biotechnology* 37 (4):349–61. doi: 10.1007/s10295-009-0680-2.

Nascimento, K. S., A. I. Cunha, K. S. Nascimento, B. S. Cavada, A. M. Azevedo, and M. R. Aires, Barros. 2012. An overview of lectins purification strategies. *Journal of Molecular Recognition: JMR* 25 (11):527–41. doi: 10.1002/jmr.2200.

Navarre, W. W., and O. Schneewind. 1999. Surface proteins of gram-positive bacteria and mechanisms of their targeting to the cell wall envelope. *Microbiology and Molecular Biology Reviews: MMBR* 63 (1):174–229. doi: 10.1128/MMBR.63.1.174-229.1999.

Ndaw, S., M. Bergaentzle, D. Aoudé-Werner, and C. Hasselmann. 2002. Enzymatic extraction procedure for the liquid chromatographic determination of niacin in foodstuffs. *Food Chemistry* 78 (1):129–34. doi.org/ (02)00:129–34. 4 doi: 10.1016/S0308-8146.

Neira, H. D., and A. E. Herr. 2017. Kinetic analysis of enzymes immobilized in porous film arrays. *Analytical Chemistry* 89 (19):10311–20. doi: 10.1021/acs.analchem.7b02075.

Nematian, T., A. Shakeri, Z. Salehi, and A. A. Saboury. 2020. Lipase immobilized on functionalized superparamagnetic few-layer graphene oxide as an efficient nanobiocatalyst for biodiesel production from *Chlorella vulgaris* bio-oil. *Biotechnology for Biofuels* 13 (1):1–15. doi: 10.1186/s13068-020-01688-x.

Nerurkar, M., M. Joshi, S. Pariti, and R. Adivarekar. 2013. Application of lipase from marine bacteria *Bacillus sonorensis* as an additive in detergent formulation. *Journal of Surfactants and Detergents* 16 (3):435–43. doi: 10.1007/s11743-012-1434-0.

Nguyen, H. H., and M. Kim. 2017. An overview of techniques in enzyme immobilization. *Applied Science and Convergence Technology* 26 (6):157–63. doi: 10.5757/ASCT.2017.26.6.157.

Nicholson, R. A., and A. G. Marangoni. 2021. Lipase-catalyzed glycerolysis extended to the conversion of a variety of edible oils into structural fats. *Current Research in Food Science* 4:163–74. doi: 10.1016/j.crfs.2021.03.005.

Nishio, T., M. Kamimura, M. Murata, Y. Terao, and K. Achiwa. 1989. Production of optically active esters and alcohols from racemic alcohols by lipase-catalyzed stereoselective transesterification in non-aqueous reaction system. *Journal of Biochemistry* 105 (4):510–2. doi: 10.1093/oxfordjournals.jbchem.a122697.

Niu, J.-F., G. Wang, and C. Tseng. 2006. Method for large-scale isolation and purification of R-phycoerythrin from red alga *Polysiphonia urceolata* Grev. *Protein Expression and Purification* 49 (1):23–31. doi: 10.1016/j.pep.2006.02.001.

Nordwald, E. M., G. S. Armstrong, and J. L. Kaar. 2014. NMR-guided rational engineering of an ionic-liquid-tolerant lipase. *ACS Catalysis* 4 (11):4057–64. doi: 10.1021/cs500978x.

Nowak, P., K. Kucharska, and M. Kamiński. 2019. Ecological and health effects of lubricant oils emitted into the environment. *International Journal of Environmental Research and Public Health* 16 (16):3002. doi: 10.3390/ijerph16163002.

Numan, M. T., and N. B. Bhosle. 2006. α Alpha-L-arabinofuranosidases: the potential applications in biotechnology. *Journal of Industrial Microbiology & Biotechnology* 33 (4):247–60. doi: 10.1007/s10295-005-0072-1.

Nyendak, M. R., D. A. Lewinsohn, and D. M. Lewinsohn. 2009. New diagnostic methods for tuberculosis. *Current Opinion in Infectious Diseases* 22 (2):3889480–174. PMCID: PMC:3889480–174.

Ohgiya, S., T. Hoshino, H. Okuyama, S. Tanaka, and K. Ishizaki. 1999. Biotechnology of enzymes from cold-adapted microorganisms. In *Biotechnological applications of cold-adapted organisms*, ed. R. Margesin, and F. Schinner, 17–34. Berlin, Heidelberg: Springer. doiorg/ doi: 10.1007/978-3-642-58607-1_2.

Okino-Delgado, C. H., D. Z. d. Prado, R. Facanali, M. M. O. Marques, A. S. Nascimento, C. J. d C. Fernandes, W. F. Zambuzzi, and L. F. Fleuri. 2017. Bioremediation of cooking oil waste using lipases from wastes. *PloS One* 12 (10):e0186246. doi: 10.1371/journal.pone.0186246.

Oliveira de Medeiros, F., C. A. V. Burkert, and S. J. Kalil. 2012. Purification of β-galactosidase by ion exchange chromatography: Elution optimization using an experimental design. *Chemical Engineering & Technology* 35 (5):911–8. doi: 10.1002/ceat.201100571.

Olusesan, A. T., L. K. Azura, F. Abubakar, N. S. A. Hamid, S. Radu, and N. Saari. 2009. Phenotypic and molecular identification of a novel thermophilic *Anoxybacillus* species: A lipase-producing bacterium isolated from a Malaysian hotspring. *World Journal of Microbiology and Biotechnology* 25 (11):1981–8. doi: 10.1007/s11274-009-0097-0.

Ong, A. L., A. H. Kamaruddin, and S. Bhatia. 2005. Current technologies for the production of (S)-ketoprofen: Process perspective. *Process Biochemistry* 40 (11):3526–35. doi: 10.1016/j.procbio.2005.03.054.

Ota, Y., T. Sawamoto, and M. Hasuo. 2000. Tributyrin specifically induces a lipase with a preference for the sn-2 position of triglyceride in *Geotrichum* sp. FO401B. *Bioscience, Biotechnology, and Biochemistry* 64 (11):2497–9. doi: 10.1271/bbb.64.2497.

Ota, Y., and K. Yamada. 1966. Lipase from *Candida paralipolytica*: Part I. Anionic surfactants as the essential activator in the systems emulsified by polyvinyl alcohol. *Agricultural and Biological Chemistry* 30 (4):351–8. doi: 10.1080/00021369.1966.10858605.

Owusu-Ansah, Y. J. 1994. Enzymes in lipid technology and cocoa butter substitutes. In *Technological advances in improved and alternative sources of lipids*, ed. B. S. Kamel, and Y. Kakuda. Boston, MA: Springer. doi: 10.1007/978-1-4615-2109-9_12.

Ozturkoglu-Budak, S., A. Wiebenga, P. A. Bron, and R. P. d. Vries. 2016. Protease and lipase activities of fungal and bacterial strains derived from an artisanal raw ewe's milk cheese. *International Journal of Food Microbiology* 237:17–27. doi: 10.1016/j.ijfoodmicro.2016.08.007.

Palekar, A. A., P. T. Vasudevan, and S. Yan. 2000. Purification of lipase: A review. *Biocatalysis and Biotransformation* 18 (3):177–200. doi: 10.3109/10242420009015244.

Palissa, H., H. von Döhren, H. Kleinkauf, H. H. Ting, and J. E. Baldwin. 1989. Beta-lactam biosynthesis in a gram-negative eubacterium: Purification and characterization of isopenicillin N synthase from *Flavobacterium* sp. strain SC 12.154. *Journal of Bacteriology* 171 (10):5720–8. doi: 10.1128/jb.171.10.5720-5728.1989.

Palomo, J. M., C. Ortiz, M. Fuentes, G. Fernandez-Lorente, J. M. Guisan, and R. Fernandez-Lafuente. 2004. Use of immobilized lipases for lipase purification via specific lipase-lipase interactions. *Journal of Chromatography. A* 1038 (1–2):267–73. doi: 10.1016/j.chroma.2004.03.058.

Paluzar, H., D. Tuncay, and H. Aydogdu. 2021. Production and characterization of lipase from *Penicillium aurantiogriseum* under solid-state fermentation using sunflower pulp. *Biocatalysis and Biotransformation* 39 (4):333–42. doi.org/:333–42. doi: 10.1080/10242422.2021.1901888.

Paraskevopoulou, V., and F. H. Falcone. 2018. Polyionic tags as enhancers of protein solubility in recombinant protein expression. *Microorganisms* 6 (2):47. doi: 10.3390/microorganisms6020047.

Park, M., E. Do, and W. H. Jung. 2013. Lipolytic enzymes involved in the virulence of human pathogenic fungi. *Mycobiology* 41 (2):67–72. doi: 10.5941/MYCO.2013.41.2.67.

Park, S. H., and H. K. Kim. 2020. Antibacterial activity of emulsions containing unsaturated fatty acid ergosterol esters synthesized by lipase-mediated transesterification. *Enzyme and Microbial Technology* 139:109581. doi: 10.1016/j.enzmictec.2020.109581.

Park, Y.-K., M. Vandermies, P. Soudier, S. Telek, S. Thomas, J.-M. Nicaud, and P. Fickers. 2019. Efficient expression vectors and host strain for the production of recombinant proteins by *Yarrowia lipolytica* in process conditions. *Microbial Cell Factories* 18 (1):1–12. doi: 10.1186/s12934-019-1218-6.

Passolunghi, S., S. Brocca, L. Cannizzaro, D. Porro, and M. Lotti. 2003. Monitoring the transport of recombinant *Candida rugosa* lipase by a green fluorescent protein-lipase fusion. *Biotechnology Letters* 25 (22):1945–8. doi: 10.1023/B:BILE.0000003991.71854.09.

Patel, N., D. Rai, S. Shahane, and U. Mishra. 2019. Lipases: Sources, production, purification, and applications. *Recent Patents on Biotechnology* 13 (1):45–56. doi: 10.2174/1872208312666181029093333.

Pathak, V. M., and Navneet. 2017. Review on the current status of polymer degradation: A microbial approach. *Bioresources and Bioprocessing* 4 (1):1–31. doi: 10.1186/s40643-017-0145-9.

Pereira, E. B., H. F. D. Castro, F. F. D. Moraes, and G. M. Zanin. 2001. Kinetic studies of lipase from *Candida rugosa*. In *Twenty-second symposium on biotechnology for fuels and chemicals*, 739–52. Totowa, NJ: Humana Press. doi: 10.1007/978-1-4612-0217-2_62.

Pereira, M. G., A. C. Vici, F. D. A. Facchini, A. P. Tristao, J. R. Cursino-Santos, P. R. Sanches, J. A. Jorge, M. de Lourdes, and T. d M. Polizeli. 2014. Screening of filamentous fungi for lipase production: *Hypocrea pseudokoningii* a new producer with a high biotechnological potential. *Biocatalysis and Biotransformation* 32 (1):74–83. doi: 10.3109/10242422.2013.873417.

Pichot, R., R. L. Watson, and I. T. Norton. 2013. Phospholipids at the interface: Current trends and challenges. *International Journal of Molecular Sciences* 14 (6):11767–94. doi: 10.3390/ijms140611767.

Pillay, V., C. M. Dangor, T. Govender, K. R. Moopanar, and N. Hurbans. 1998. Ionotropic gelation: Encapsulation of indomethacin in calcium alginate gel discs. *Journal of Microencapsulation* 15 (2):215–26. doi: 10.3109/02652049809006851.

Pinheiro, B. B., N. S. Rios, E. R. Aguado, R. Fernandez-Lafuente, T. M. Freire, P. B. A. Fechine, J. C. S. Dos Santos, and L. R. B. Goncalves. 2019. Chitosan activated with divinyl sulfone: A new heterofunctional support for enzyme immobilization. Application in the immobilization of lipase B from *Candida antarctica*. *International Journal of Biological Macromolecules* 130:798–809. doi: 10.1016/j.ijbiomac.2019.02.145.

Pinheiro, M. P., N. S. Rios, T. d. S. Fonseca, F. d. A. Bezerra, E. Rodríguez-Castellón, R. Fernandez-Lafuente, M. C. de Mattos, J. C. S. Dos Santos, and L. R. B. Gonçalves. 2018. Kinetic resolution of drug intermediates catalyzed by lipase B from *Candida antarctica* immobilized on immobead-350. *Biotechnology Progress* 34 (4):878–89. doi: 10.1002/btpr.2630.

Pulido, I. Y., E. Prieto, G. P. Pieffet, L. Méndez, and C. A. Jiménez-Junca. 2020. Functional heterologous expression of mature lipase LipA from *Pseudomonas aeruginosa* PSA01 in *Escherichia coli* SHuffle and BL21 (DE3): Effect of the expression host on thermal stability and solvent tolerance of the enzyme produced. *International Journal of Molecular Sciences* 21 (11):3925. doi: 10.3390/ijms21113925.

Putra, L., G. H. Natadiputri, A. Meryandini, and A. Suwanto. 2019. Isolation, cloning and co-expression of lipase and foldase genes of burkholderia territorii GP3 from Mount Papandayan soil. *Journal of Microbiology and Biotechnology* 29 (6):944–51. doi: 10.4014/jmb.1812.12013.

Qin, L.-N., F.-R. Cai, X.-R. Dong, Z.-B. Huang, Y. Tao, J.-Z. Huang, and Z.-Y. Dong. 2012. Improved production of heterologous lipase in *Trichoderma reesei* by RNAi mediated gene silencing of an endogenic highly expressed gene. *Bioresource Technology* 109:116–22. doi: 10.1016/j.biortech.2012.01.013.

Qin, L., X. Jiang, Z. Dong, J. Huang, and X. Chen. 2018. Identification of two integration sites in favor of transgene expression in *Trichoderma reesei*. *Biotechnology for Biofuels* 11 (1):1–15. doi: 10.1186/s13068-018-1139-3.

Raclot, T., C. Holm, and D. Langin. 2001. Fatty acid specificity of hormone-sensitive lipase: Implication in the selective hydrolysis of triacylglycerols. *Journal of Lipid Research* 42 (12):2049–57. doi: 10.1016/S0022-2275. (20)31534-0

Rafiee, F., and M. Rezaee. 2021. Different strategies for the lipase immobilization on the chitosan based supports and their applications . *International Journal of Biological Macromolecules* 179:170–95. doi: 10.1016/j.ijbiomac.2021.02.198.

Raftari, M., S. Ghafourian, N. Sadeghifard, F. Abu Bakar, N. Saari, and Z. Sekawi. 2012. Overexpression of recombinant lipase from *Burkholderia cepacia* in *Escherichia coli*. *European Journal of Inflammation* 10 (3):365–9. doi: 10.1177/1721727X1201000312.

Rajendran, A., A. Palanisamy, and V. Thangavelu. 2009. Lipase catalyzed ester synthesis for food processing industries. *Brazilian Archives of Biology and Technology* 52 (1):207–19. doi: 10.1590/S1516-89132009000100026.

Rani, S., and S. Jagtap. 2019. Acceleration of Swiss cheese ripening by microbial lipase without affecting its quality characteristics. *Journal of Food Science and Technology* 56 (1):497–506. doi: 10.1007/s13197-018-3482-6.

Ranjan, M. T., A. R. Byreddy, M. Puri, C. Barrow, and N. M. Rao. 2016. Selective enrichment of omega-3 fatty acids in oils by phos-

pholipase A1. *PloS One* 11 (3):e0151370. doi: 10.1371/journal.pone.0151370.

Rantasalo, A., M. Vitikainen, T. Paasikallio, J. Jäntti, C. P. Landowski, and D. Mojzita. 2019. Novel genetic tools that enable highly pure protein production in *Trichoderma reesei*. *Scientific Reports* 9 (1):1–12. doi: 10.1038/s41598-019-41573-8.

Rasamiravaka, T., Q. Labtani, P. Duez, and M. E. Jaziri. 2015. The formation of biofilms by *Pseudomonas aeruginosa*: A review of the natural and synthetic compounds interfering with control mechanisms. *BioMed Research International* 2015:1–17. Article ID 759348 | :1–17. doi: 10.1155/2015/759348.

Rasmussen-Ivey, C. R., M. J. Figueras, D. McGarey, and M. R. Liles. 2016. Virulence factors of *Aeromonas hydrophila*: In the wake of reclassification. *Frontiers in Microbiology* 7:1337. doi: 10.3389/fmicb.2016.01337.

Raveendran, S., B. Parameswaran, S. B. Ummalyma, A. Abraham, A. K. Mathew, A. Madhavan, S. Rebello, and A. Pandey. 2018. Applications of microbial enzymes in food industry. *Food Technology and Biotechnology* 56 (1):16–30. doi: 10.17113/ftb.56.01.18.5491.

Reetz, M. T. 2002. Lipases as practical biocatalysts. *Current Opinion in Chemical Biology* 6 (2):145–50. doi: 10.1016/S1367-5931(02)00297-1.

Rengachari, S., G. A. Bezerra, L. Riegler-Berket, C. C. Gruber, C. Sturm, U. Taschler, A. Boeszoermenyi, I. Dreveny, R. Zimmermann, K. Gruber, et al. 2012. The structure of monoacylglycerol lipase from *Bacillus* sp. H257 reveals unexpected conservation of the cap architecture between bacterial and human enzymes. *Biochimica et Biophysica Acta* 1821 (7):1012–21. doi: 10.1016/j.bbalip.2012.04.006.

Resina, D., O. Cos, P. Ferrer, and F. Valero. 2005. Developing high cell density fed-batch cultivation strategies for heterologous protein production in Pichia pastoris using the nitrogen source-regulated FLD1 Promoter. *Biotechnology and Bioengineering* 91 (6):760–7. doi: 10.1002/bit.20545.

Ribeiro, W. F., T. M. G. Selva, I. C. Lopes, E. C. S. Coelho, S. G. Lemos, F. C. d. Abreu, V. B. d. Nascimento, and M. C. U. d. Araújo. 2011. Electroanalytical determination of carbendazim by square wave adsorptive stripping voltammetry with multiwalled carbon nanotubes modified electrode. *Analytical Methods* 3 (5):1202–6. doi: 10.1039/c0ay00723d.

Richmond, J. A., T. L. Dunning, and P. V. Desmond. 2007. Health professionals' attitudes toward caring for people with hepatitis C. *Journal of Viral Hepatitis* 14 (9):624–32. doi: 10.1111/j.1365-2893.2007.00849.x.

Robert, J. M., M. O. Betancur, A. C. O. Machado, A. Arruda, V. C. B. Reis, R. V. Almeida, F. A. G. Torres, P. F. Alegre, F. Valero, and D. M. G. Freire. 2019. Increase of *Candida antarctica* lipase B production under PGK promoter in *Pichia pastoris*: Effect of multicopies. *Brazilian Journal of Microbiology: [Publication of the Brazilian Society for Microbiology]* 50 (2):405–13. doi: 10.1007/s42770-019-00056-8.

Robinson, P. K. 2015. Correction: Enzymes: Principles and biotechnological applications. *Essays in Biochemistry* 59:1–75. doi: 10.1042/bse0590001.

Robles-Medina, A., P. A. González-Moreno, L. Esteban-Cerdán, and E. Molina-Grima. 2009. Biocatalysis: Towards ever greener biodiesel production. *Biotechnology Advances* 27 (4):398–408. doi: 10.1016/j.biotechadv.2008.10.008.

Rodrigues, R. C., and M. A. Z. Ayub. 2011. Effects of the combined use of *Thermomyces lanuginosus* and *Rhizomucor miehei* lipases for the transesterification and hydrolysis of soybean oil. *Process Biochemistry* 46 (3):682–8. doi: 10.1016/j.procbio.2010.11.013.

Rodriguez-Abetxuko, A., D. Sánchez-deAlcázar, P. Muñumer, and A. Beloqui. 2020. Tunable polymeric scaffolds for enzyme immobilization. *Frontiers in Bioengineering and Biotechnology* 8:830. doi: 10.3389/fbioe.2020.00830.

Rogalska, E., C. Cudrey, F. Ferrato, and R. Verger. 1993. Stereoselective hydrolysis of triglycerides by animal and microbial lipases. *Chirality* 5 (1):24–30. doi: 10.1002/chir.530050106.

Rosano, G. L., and E. A. Ceccarelli. 2014. Recombinant protein expression in *Escherichia coli:* Advances and challenges. *Frontiers in Microbiology* 5:172. doi: 10.3389/fmicb.2014.00172.

Rosenthal, K., and S. Lütz. 2018. Recent developments and challenges of biocatalytic processes in the pharmaceutical industry. *Current Opinion in Green and Sustainable Chemistry* 11:58–64. doi: 10.1016/j.cogsc.2018.03.015.

Royter, M., M. Schmidt, C. Elend, H. Höbenreich, T. Schäfer, U. T. Bornscheuer, and G. Antranikian. 2009. Thermostable lipases from the extreme thermophilic anaerobic bacteria *Thermoanaerobacter thermohydrosulfuricus* SOL1 and *Caldanaerobacter subterraneus* subsp. tengcongensis. *Extremophiles: Life under Extreme Conditions* 13 (5):769–83. doi: 10.1007/s00792-009-0265-z.

Rubio, R., A. Jofré, B. Martín, T. Aymerich, and M. Garriga. 2014. Characterization of lactic acid bacteria isolated from infant faeces as potential probiotic starter cultures for fermented sausages. *Food Microbiology* 38 (2014):303–11. doi: 10.1016/j.fm.2013.07.015.

Saeui, C. T., E. Urias, L. Liu, M. P. Mathew, and K. J. Yarema. 2015. Metabolic glycoengineering bacteria for therapeutic, recombinant protein, and metabolite production applications. *Glycoconjugate Journal* 32 (7):425–41. doi: 10.1007/s10719-015-9583-9.

Sağiroğlu, A., A. Kilinç, and A. Elefoncu. 2004. Preparation and properties of lipases immobilized on different supports. *Artificial Cells, Blood Substitutes, and Immobilization Biotechnology* 32 (4):625–36. doi: 10.1081/bio-200039656.

Sahin, N., C. C. Akoh, and A. Karaali. 2005. Lipase-catalyzed acidolysis of tripalmitin with hazelnut oil fatty acids and stearic acid to produce human milk fat substitutes. *Journal of Agricultural and Food Chemistry* 53 (14):5779–83. doi: 10.1021/jf050465e.

Sakai, S., Y. Liu, T. Yamaguchi, R. Watanabe, M. Kawabe, and K. Kawakami. 2010. Immobilization of *Pseudomonas cepacia* lipase onto electrospun polyacrylonitrile fibers through physical adsorption and application to transesterification in nonaqueous solvent. *Biotechnology Letters* 32 (8):1059–62. doi: 10.1007/s10529-010-0279-8.

Sales, J. C. S., A. M. d. Castro, B. D. Ribeiro, and M. A. Z. Coelho. 2020. Supplementation of watermelon peels as an enhancer of lipase and esterase production by *Yarrowia lipolytica* in solid-state fermentation and their potential use as biocatalysts in poly (ethylene terephthalate) (PET) depolymerization reactions. *Biocatalysis and Biotransformation* 38 (6):457–68. doi: 10.1080/10242422.2020.1782387.

Salihu, A., and M. Zahangir Alam. 2015. Solvent tolerant lipases: A review. *Process Biochemistry* 50 (1):86–96. doi: 10.1016/j.procbio.2014.10.019.

Salleh, A. B., Siti, M. Baharuddin, Raja, N. Z. R. A. Rahman, T. C. Leow, M. Basri, and S. N. Oslan. 2020. A host-vector system for the expression of a thermostable bacterial lipase in a locally isolated *Meyerozyma guilliermondii* SMB. *Microorganisms* 8 (11):1738. doi: 10.3390/microorganisms8111738.

Salvi, H. M., M. P. Kamble, and G. D. Yadav. 2018. Synthesis of geraniol esters in a continuous-flow packed-bed reactor of immobilized lipase: Optimization of process parameters and kinetic modeling. *Applied Biochemistry and Biotechnology* 184 (2):630–43. doi: 10.1007/s12010-017-2572-7.

Salwoom, L., R. N. Z. R. Abd Rahman, A. B. Salleh, P. Convey, D. Pearce, and M. S. M. Ali. 2019. Isolation, characterization, and lipase production of a cold-adapted bacterial strain *Pseudomonas* sp. LSK25 isolated from Signy Island, Antarctica. *Molecules* 24 (4):715. doi: 10.3390/molecules24040715.

Samiey, B., C.-H. Cheng, and J. Wu. 2014. Organic-inorganic hybrid polymers as adsorbents for removal of heavy metal ions from solutions: A review. *Materials (Basel, Switzerland)* 7 (2):673–726. doi: 10.3390/ma7020673.

Samuel, P., A. K. P. Vadhana, R. Kamatchi, A. Antony, and S. Meenakshisundaram. 2013. Effect of molecular chaperones on the expression of *Candida antarctica* lipase B in *Pichia pastoris*. *Microbiological Research* 168 (10):615–20. doi: 10.1016/j.micres.2013.06.007.

Sanchez, S., and A. L. Demain. 2011. Enzymes and bioconversions of industrial, pharmaceutical, and biotechnological significance. *Organic Process Research & Development* 15 (1):224–30. doi: 10.1021/op100302x.

Sánchez-Hernández, S., A. Esteban-Muñoz, R. Giménez-Martínez, M. J. Aguilar-Cordero, B. Miralles-Buraglia, and M. Olalla-Herrera.

2019. A comparison of changes in the fatty acid profile of human milk of Spanish lactating women during the first month of lactation using gas chromatography-mass spectrometry. A comparison with infant formulas. *Nutrients* 11 (12):3055. doi: 10.3390/nu11123055.

Sankaran, R., P. L. Show, and J.-S. Chang. 2016. Biodiesel production using immobilized lipase: Feasibility and challenges. *Biofuels, Bioproducts and Biorefining* 10 (6):896–916. doi: 10.1002/bbb.1719.

Santilli, A. A., A. C. Scotese, R. F. Bauer, and S. C. Bell. 1987. 2-Oxo-1,8-naphthyridine-3-carboxylic acid derivatives with potent gastric antisecretory properties. *Journal of Medicinal Chemistry* 30 (12):2270–7. doi: 10.1021/jm00395a015.

Sarkar, P., S. Yamasaki, S. Basak, A. Bera, and P. K. Bag. 2012. Purification and characterization of a new alkali-thermostable lipase from *Staphylococcus aureus* isolated from *Arachis hypogaea* rhizosphere. *Process Biochemistry* 47 (5):858–66. doi: 10.1016/j.procbio.2012.02.023.

Sarmah, N., D. Revathi, G. Sheelu, K. Y. Rani, S. Sridhar, V. Mehtab, and C. Sumana. 2018. Recent advances on sources and industrial applications of lipases. *Biotechnology Progress* 34 (1):5–28. doi: 10.1002/btpr.2581.

Sato, H., T. Watanabe, Y. Murata, A. Ohtake, M. Nakamura, C. Aizawa, H. Saito, and N. Maehara. 1999. New exfoliative toxin produced by a plasmid-carrying strain of *Staphylococcus hyicus*. *Infection and Immunity* 67 (8):4014–8. doi: 10.1128/IAI.67.8.4014-4018.1999.

Saxena, R. K., A. Sheoran, B. Giri, and W. S. Davidson. 2003. Purification strategies for microbial lipases. *Journal of Microbiological Methods* 52 (1):1–18. doi: 10.1016/S0167-7012(02)00161-6.

Schirone, M., R. Tofalo, P. Visciano, A. Corsetti, and G. Suzzi. 2012. Biogenic amines in Italian Pecorino cheese. *Frontiers in Microbiology* 3:171. doi: 10.3389/fmicb.2012.00171.

Schmid, R. D., and R. Verger. 1998. Lipases: Interfacial enzymes with attractive applications. *Angewandte Chemie International Edition* 37 (12):1608–33. doi: 10.1002/(SICI)1521-3773(19980703)37:12<1608::AID-ANIE1608>3.0.CO;2-V.

Schoemaker, H. E., D. Mink, and M. G. Wubbolts. 2003. Dispelling the myths-biocatalysis in industrial synthesis. *Science (New York, NY)* 299 (5613):1694–7. doi: 10.1126/science.1079237.

Schoffelen, S., and J. C. M V. Hest. 2013. Chemical approaches for the construction of multi-enzyme reaction systems. *Current Opinion in Structural Biology* 23 (4):613–21. doi: 10.1016/j.sbi.2013.06.010.

Schomburg, I., A. Chang, and D. Schomburg. 2014. Standardization in enzymology—Data integration in the world′ s enzyme information system BRENDA. *Perspectives in Science* 1 (1–6):15–23. doi: 10.1016/j.pisc.2014.02.002.

Seitz, E. W. 1974. Industrial application of microbial lipases: A review. *Journal of the American Oil Chemists' Society* 51 (2):12–6. doi: 10.1007/BF02545206.

Sen, K., and M. Rodgers. 2004. Distribution of six virulence factors in *Aeromonas* species isolated from US drinking water utilities: A PCR identification. *Journal of Applied Microbiology* 97 (5):1077–86. doi: 10.1111/j.1365-2672.2004.02398.x.

Senbua, W., J. Mearnchu, and J. Wichitwechkarn. 2020. Easy-to-use and reliable absorbance-based MPH-GST biosensor for the detection of methyl parathion pesticides. *Biotechnology Reports (Amsterdam, Netherlands)* 27:e00495. doi: 10.1016/j.btre.2020.e00495.

Sha, C., X.-W. Yu, N.-X. Lin, M. Zhang, and Y. Xu. 2013. Enhancement of lipase r27RCL production in *Pichia pastoris* by regulating gene dosage and co-expression with chaperone protein disulfide isomerase. *Enzyme and Microbial Technology* 53 (6–7):438–43. doi: 10.1016/j.enzmictec.2013.09.009.

Shao, H., X. Hu, L. Sun, and W. Zhou. 2019. Gene cloning, expression in *E. coli*, and in vitro refolding of a lipase from Proteus sp. NH 2-2 and its application for biodiesel production. *Biotechnology Letters* 41 (1):159–69. doi: 10.1007/s10529-018-2625-1.

Sharma, R., Y. Chisti, and U. C. Banerjee. 2001. Production, purification, characterization, and applications of lipases. *Biotechnology Advances* 19 (8):627–62. doi: 10.1016/S0734-9750(01)00086-6.

Sharma, A., K. S. Thatai, T. Kuthiala, G. Singh, and S. K. Arya. 2021. Employment of polysaccharides in enzyme immobilization. *Reactive and Functional Polymers* 167 (2021):105005. doi: 10.1016/j.reactfunctpolym.2021.105005.

Sheldon, R. A. 2007. Cross-linked enzyme aggregates (CLEA®s): stable and recyclable biocatalysts. *Biochemical Society Transactions* 35 (6):1583–7. doi:10.1042/BST0351583.

Sheldon, R. A., and S. van Pelt. 2013. Enzyme immobilisation in biocatalysis: why, what and how. *Chemical Society Reviews* 42 (15):6223–35. doi: 10.1039/C3CS60075K.

Shockey, J., D. Chapital, S. Gidda, C. Mason, G. Davis, K. T. Klasson, H. Cao, R. Mullen, and J. Dyer. 2011. Expression of a lipid-inducible, self-regulating form of *Yarrowia lipolytica* lipase LIP2 in *Saccharomyces cerevisiae*. *Applied Microbiology and Biotechnology* 92 (6):1207–17. doi: 10.1007/s00253-011-3505-y.

Siebenhaller, S., J. Gentes, A. Infantes, C. Muhle-Goll, F. Kirschhöfer, G. Brenner-Weiß, O. Katrin, and S. Christoph. 2018. Lipase-catalyzed synthesis of sugar esters in honey and agave syrup. *Frontiers in Chemistry* 6 (2018):24. doi: 10.3389/fchem.2018.00024.

Siebenhaller, S., T. Hajek, C. Muhle-Goll, M. Himmelsbach, B. Luy, F. Kirschhöfer, G. Brenner-Weiß, T. Hahn, S. Zibek, and C. Syldatk. 2017. Beechwood carbohydrates for enzymatic synthesis of sustainable glycolipids. *Bioresources and Bioprocessing* 4 (1):1–9. doi: 10.1186/s40643-017-0155-7.

Sigurdardóttir, S. B., J. Lehmann, S. Ovtar, J. Grivel, M. D. Negra, A. Kaiser, and M. Pinelo. 2018. Enzyme immobilization on inorganic surfaces for membrane reactor applications: Mass transfer challenges, enzyme leakage and reuse of materials. *Advanced Synthesis & Catalysis* 360 (14):2578–607. doi: 10.1002/adsc.201800307.

Silva, J. E. S., and P. C. Jesus. 2003. Evaluation of the catalytic activity of lipases immobilized on chrysotile for esterification. *Anais da Academia Brasileira de Ciencias* 75 (2):157–62. 75: 157-162. doi: 10.1590/s0001-37652003000200003.

Singh, A. K., and M. Mukhopadhyay. 2012. Overview of fungal lipase: A review. *Applied Biochemistry and Biotechnology* 166 (2):486–520. doi: 10.1007/s12010-011-9444-3.

Singh, R., A. K. Upadhyay, P. Chandra, and D. P. Singh. 2018. Sodium chloride incites reactive oxygen species in green algae *Chlorococcum humicola* and *Chlorella vulgaris*: Implication on lipid synthesis, mineral nutrients and antioxidant system. *Bioresource Technology* 270:489–97. doi: 10.1016/j.biortech.2018.09.065.

Sirisha, V. L., A. Jain, and A. Jain. 2016. Enzyme immobilization: An overview on methods, support material, and applications of immobilized enzymes. *Advances in Food and Nutrition Research* 79:179–211. doi: 10.1016/bs.afnr.2016.07.004.

Soares, C. M. F., H. F. D. Castro, F. F. De Moraes, and G. M. Zanin. 1999. Characterization and utilization of *Candida rugosa* lipase immobilized on controlled pore silica. In *Twentieth symposium on biotechnology for fuels and chemicals. Applied biochemistry and biotechnology*, ed. B. H. Davison and M. Finkelstein. Totowa, NJ: Humana Press. doi: 10.1007/978-1-4612-1604-9_68.

Sobczak, A. I. S., C. A. Blindauer, and A. J. Stewart. 2019. Changes in plasma-free fatty acids associated with type-2 diabetes. *Nutrients* 11 (9):2022. doi: 10.3390/nu11092022.

Soleymani, S., H. Alizadeh, H. Mohammadian, E. Rabbani, F. Moazen, H. MirMohammad Sadeghi, Z. S. Shariat, Z. Etemadifar, and M. Rabbani. 2017. Efficient media for high lipase production: One variable at a time approach. *Avicenna Journal of medical biotechnology* 9 (2):82. PMCID: PMC5410133

Son, M., Y. Moon, M. J. Oh, S. B. Han, K. H. Park, J.-G. Kim, and J. H. Ahn. 2012. Lipase and protease double-deletion mutant of *Pseudomonas fluorescens* suitable for extracellular protein production. *Applied and Environmental Microbiology* 78 (23):8454–62. doi: 10.1128/AEM.02476-12.

Song, J., J. Park, J. Jung, C. Lee, S. Y. Gim, H. Ka, B. Yi, M.-J. Kim, C-i. Kim, and J. Lee. 2015. Analysis of trans fat in edible oils with the cooking process. *Toxicological Research* 31 (3):307–12. doi: 10.5487/TR.2015.31.3.307.

Soumanou, M. M., M. Pérignon, and P. Villeneuve. 2013. Lipase-catalyzed interesterification reactions for human milk fat substitute's production: A review. *European Journal of Lipid Science and Technology* 115 (3):270–85. doi: 10.1002/ejlt.201200084.

Spahn, C., and S. D. Minteer. 2008. Enzyme immobilization in biotechnology. *Recent Patents on Engineering* 2 (3):195–200. doi: 10.2174/187221208786306333.

Stadler, P., A. Kovac, L. Haalck, F. Spener, and F. Paltauf. 1995. Stereoselectivity of Microbial lipases. The substitution at position sn-2 of triacylglycerol analogs influences the stereoselectivity of different microbial lipases. *European Journal of Biochemistry* 227 (1–2):335–43. doi: 10.1111/j.1432-1033.1995.tb20394.x.

Stanhope, K. L. 2012. Role of fructose-containing sugars in the epidemics of obesity and metabolic syndrome. *Annual Review of Medicine* 63:329–43. doi: 10.1146/annurev-med-042010-113026.

Stauch, B., S. J. Fisher, and M. Cianci. 2015. Open and closed states of *Candida antarctica* lipase B: Protonation and the mechanism of interfacial activation1. *Journal of Lipid Research* 56 (12):2348–58. doi: 10.1194/jlr.M063388.

Stefanovic, E., K. N. Kilcawley, C. Roces, M. C. Rea, M. O'Sullivan, J. J. Sheehan, and O. McAuliffe. 2018. Evaluation of the potential of *Lactobacillus paracasei* adjuncts for flavor compounds development and diversification in short-aged cheddar cheese. *Frontiers in Microbiology* 9 (2018):1506. doi: 10.3389/fmicb.2018.01506.

Stehr, F., A. Felk, A. Gácser, M. Kretschmar, B. Mähnss, K. Neuber, B. Hube, and W. Schäfer. 2004. Expression analysis of the *Candida albicans* lipase gene family during experimental infections and in patient samples. *FEMS Yeast Research* 4 (4–5):401–8. doi: 10.1016/S1567-1356(03)00205-8.

Stehr, F., M. Kretschmar, C. Kröger, B. Hube, and W. Schäfer. 2003. Microbial lipases as virulence factors. *Journal of Molecular Catalysis B: Enzymatic* 22 (5–6):347–55. doi: 10.1016/S1381-1177(03)00049-3.

Stephanopoulos, G. 2007. Challenges in engineering microbes for biofuels production. *Science (New York, NY)* 315 (5813):801–4. doi: 10.1126/science.1139612.

Sugahara, V. H., and G. d S. Varéa. 2014. Immobilization of *Beauveria bassiana* lipase on silica gel by physical adsorption. *Brazilian Archives of Biology and Technology* 57 (6):842–50. doi: 10.1590/S1516-8913201401358.

Sugo, K., and T. Okuyama. 2018. Hydroxyapatite chromatographic procedures for phospholipids. *Separation Science and Technology* 53 (2):389–96. doi: 10.1080/01496395.2017.1380047.

Sulaiman, S., M. N. Mokhtar, M. N. Naim, A. S. Baharuddin, and A. Sulaiman. 2015. A review: Potential usage of cellulose nanofibers (CNF) for enzyme immobilization via covalent interactions. *Applied Biochemistry and Biotechnology* 175 (4):1817–42. doi: 10.1007/s12010-014-1417-x.

Sultan, A., and P. M. Sokolove. 2001. Free fatty acid effects on mitochondrial permeability: An overview. *Archives of Biochemistry and Biophysics* 386 (1):52–61. doi: 10.1006/abbi.2000.2195.

Sunitha, S., S. Kanjilal, P. S. Reddy, and R. B. N. Prasad. 2007. Ionic liquids as a reaction medium for lipase-catalyzed methanolysis of sunflower oil. *Biotechnology Letters* 29 (12):1881–5. doi: 10.1007/s10529-007-9471-x.

Svendsen, A., I. G. Clausen, S. A. Patkar, K. Borch, and M. Thellersen. 1997. Protein engineering of microbial lipases of industrial interest. *Methods in Enzymology* 284 (1997):317–40. doi: 10.1016/S0076-6879.(97)84021-9

Tahoun, M. K., and H. A. Ali. 1986. Specificity and glyceride synthesis by mycelial lipases of *Rhizopus delemar*. *Enzyme and Microbial Technology* 8 (7):429–32. doi.org/ (86)90152-3 doi: 10.1016/0141-0229.

Taipa, M. A., M. R. Aires-Barros, and J. M. S. Cabral. 1992. Purification of lipases. *Journal of Biotechnology* 26 (2–3):111–42. doi.org/10.1016/0168-1656 (92)90001-P doi: 10.1016/0168-1656(92)90001-P.

Tamalampudi, S., S. Hama, T. Tanino, M. R. Talukder, A. Kondo, and H. Fukuda. 2007. Immobilized recombinant *Aspergillus oryzae* expressing heterologous lipase: An efficient whole-cell biocatalyst for enantioselective transesterification in non-aqueous medium. *Journal of Molecular Catalysis B: Enzymatic* 48 (1–2):33–7. doi: 10.1016/j.molcatb.2007.05.007.

Tavares, M., M. Kozak, A. Balola, and I. Sá-Correia. 2020. *Burkholderia cepacia* complex bacteria: A feared contamination risk in water-based pharmaceutical products. *Clinical Microbiology Reviews* 33 (3):e00139-19. doi: 10.1128/CMR.00139-19.

Theil, F., and F. Björkling. 1993. Specificity of *Candida antarctica* lipase B (SP 435) in the presence of lipase A in a double enantioselective transesterification. *Biotechnology Letters* 15 (6):605–8. doi: 10.1007/BF00138549.

Theron, C. W., M. Vandermies, S. Telek, S. Steels, and P. Fickers. 2020. Comprehensive comparison of *Yarrowia lipolytica* and *Pichia pastoris* for production of *Candida antarctica* lipase B. *Scientific Reports* 10 (1):1–9. doi: 10.1038/s41598-020-58683-3.

Tischer, W., and F. Wedekind. 1999. Immobilized enzymes: Methods and applications. In *Biocatalysis - From discovery to application. Topics in current chemistry*, ed. W. D. Fessner, vol 200. Berlin, Heidelberg: Springer. doi: 10.1007/3-540-68116-7_4.

Tokmakov, A. A., A. Kurotani, T. Takagi, M. Toyama, M. Shirouzu, Y. Fukami, and S. Yokoyama. 2012. Multiple post-translational modifications affect heterologous protein synthesis. *The Journal of Biological Chemistry* 287 (32):27106–16. doi: 10.1074/jbc.M112.366351.

Toldrá, F., L. Mora, and M. Reig. 2016. New insights into meat by-product utilization. *Meat Science* 120 (2016):54–9. doi: 10.1016/j.meatsci.2016.04.021.

Torbeck, L., D. Raccasi, D. E. Guilfoyle, R. L. Friedman, and D. Hussong. 2011. *Burkholderia cepacia*: This decision is overdue. *PDA Journal of Pharmaceutical Science and Technology* 65 (5):535–43. doi: 10.5731/pdajpst.2011.00793.

Toth, R., A. Toth, C. Vagvolgyi, and A. Gacser. 2017. *Candida parapsilosis* secreted lipase as an important virulence factor. *Current Protein & Peptide Science* 18 (10):1043–9. doi: 10.2174/1389203717666160813163054.

Trassaert, M., M. Vandermies, F. Carly, O. Denies, S. Thomas, P. Fickers, and J. Nicaud. 2017. New inducible promoter for gene expression and synthetic biology in *Yarrowia lipolytica*. *Microbial Cell Factories* 16 (1):1–17. doi: 10.1186/s12934-017-0755-0.

Treacy, J., A. Williams, R. Bais, K. Willson, C. Worthley, J. Reece, J. Bessell, and D. Thomas. 2001. Evaluation of amylase and lipase in the diagnosis of acute pancreatitis. *ANZ Journal of Surgery* 71 (10):577–82. doi: 10.1046/j.1445-2197.2001.02220.x.

Trivedi, U. B., D. Lakshminarayana, I. L. Kothari, N. G. Patel, H. N. Kapse, K. K. Makhija, P. B. Patel, and C. J. Panchal. 2009. Potentiometric biosensor for urea determination in milk. *Sensors and Actuators B: Chemical* 140 (1):260–6. doi: 10.1016/j.snb.2009.04.022.

Turan, H., and İ. Erkoyuncu. 2012. Salting technology in fish processing. *Progress in Food Preservation*, ed. R. Bhat, Abd. K. Alias, G. Paliyath, 297–313. doi: 10.1002/9781119962045.ch14.

Tyburczy, C., M. M. Mossoba, and J. I. Rader. 2013. Determination of trans fat in edible oils: Current official methods and overview of recent developments. *Analytical and Bioanalytical Chemistry* 405 (17):5759–72. doi: 10.1007/s00216-013-7005-z.

Ueland, P. M., H. Refsum, S. P. Stabler, M. R. Malinow, A. Andersson, and R. H. Allen. 1993. Total homocysteine in plasma or serum: Methods and clinical applications. *Clinical Chemistry* 39 (9):1764–79. doi: 10.1093/clinchem/39.9.1764.

Ugur, A., and R. Boran. 2014. Production and characterization of a cold-active and n-hexane activated lipase from a newly isolated *Serratia grimesii* RB06-22. *Biocatalysis and Biotransformation* 32 (4):222–30. doi: 10.3109/10242422.2014.934684.

Ujiie, A., H. Nakano, and Y. Iwasaki. 2016. Extracellular production of *Pseudozyma* (Candida) *antarctica* lipase B with genuine primary sequence in recombinant *Escherichia coli*. *Journal of Bioscience and Bioengineering* 121 (3):303–9. doi: 10.1016/j.jbiosc.2015.07.001.

Unni, K. N., P. Priji, S. Sajith, P. A. Faisal, and S. Benjamin. 2016. *Pseudomonas aeruginosa* strain BUP2, a novel bacterium inhabiting the rumen of Malabari goat, produces an efficient lipase. *Biologia* 71 (4):378–87. doi: 10.1515/biolog-2016-0057.

Urban, A., M. Leipelt, T. Eggert, and K. Jaeger. 2001. DsbA and DsbC affect extracellular enzyme formation in *Pseudomonas aeruginosa*. *Journal of Bacteriology* 183 (2):587–96. doi: 10.1128/JB.183.2.587-596.2001.

Uthoff, S., D. Bröker, and A. Steinbüchel. 2009. Current state and perspectives of producing biodiesel-like compounds by biotechnology. *Microbial Biotechnology* 2 (5):551–65. doi: 10.1111/j.1751-7915.2009.00139.x.

Vale, W., J. Rivier, J. Vaughan, R. McClintock, A. Corrigan, W. Woo, D. Karr, and J. Spiess. 1986. Purification and characterization of an FSH releasing protein from porcine ovarian follicular fluid. *Nature* 321 (6072):776–9. doi: 10.1038/321776a0.

Valero, F. 2012. Heterologous expression systems for lipases: A review. In *Lipases and phospholipases. Methods in molecular biology (methods and protocols)*, ed. G. Sandoval, vol 861. New York: Humana Press. doi: 10.1007/978-1-61779-600-5_11.

Van Geest, M., and J. S. Lolkema. 2000. Membrane topology and insertion of membrane proteins: Search for topogenic signals. *Microbiology and Molecular Biology Reviews: MMBR* 64 (1):13–33. doi: 10.1128/MMBR.64.1.13-33.2000.

van Hoogevest, P., and A. Wendel. 2014. The use of natural and synthetic phospholipids as pharmaceutical excipients. *European Journal of Lipid Science and Technology: EJLST* 116 (9):1088–107. doi: 10.1002/ejlt.201400219.

Vasil, M. L. 1986. *Pseudomonas aeruginosa*: Biology, mechanisms of virulence, epidemiology. *The Journal of Pediatrics* 108 (5 Pt 2):800–5. doi: 10.1016/S0022-3476(86)80748-X.

Ventura, S. P. M., and J. A. P. Coutinho. 2016. Lipase production and purification from fermentation broth using ionic liquids. In *Ionic liquids in lipid processing and analysis*, 59–97. USA: Academic Press and AOCS Press. doi: 10.1016/B978-1-63067-047-4.00003-9.

Vieira Gomes, A. M., T. S. Carmo, L. S. Carvalho, F. M. Bahia, and N. S. Parachin. 2018. Comparison of yeasts as hosts for recombinant protein production. *Microorganisms* 6 (2):38. doi: 10.3390/microorganisms6020038.

Villarino, C. B. J., V. Jayasena, R. Coorey, S. Chakrabarti-Bell, and S. K. Johnson. 2016. Nutritional, health, and technological functionality of lupin flour addition to bread and other baked products: Benefits and challenges. *Critical Reviews in Food Science and Nutrition* 56 (5):835–57. doi: 10.1080/10408398.2013.814044.

Wang, Y., C. Li, Y. Zhao, L. Li, X. Yang, Y. Wu, S. Chen, J. Cen, S. Yang, and D. Yang. 2020. Novel insight into the formation mechanism of volatile flavor in Chinese fish sauce (Yu-lu) based on molecular sensory and metagenomics analyses. *Food Chemistry* 323:126839. doi: 10.1016/j.foodchem.2020.126839.

Wang, F., T. T. Nie, L. L. Shao, and Z. Cui. 2014. Comparison of physical and covalent immobilization of lipase from *Candida antarctica* on polyamine microspheres of alkylamine matrix. *Biocatalysis and Biotransformation* 32 (5–6):314–26. doi: 10.3109/10242422.2014.977266.

Wang, Z.-G., L.-S. Wan, Z.-M. Liu, X.-J. Huang, and Z.-K. Xu. 2009. Enzyme immobilization on electrospun polymer nanofibers: An overview. *Journal of Molecular Catalysis B: Enzymatic* 56 (4):189–95. doi: 10.1016/j.molcatb.2008.05.005.

Wang, J., Z. Wu, T. Zhang, Y. Wang, and B. Yang. 2019. High-level expression of *Thermomyces dupontii* thermophilic lipase in *Pichia pastoris* via combined strategies. *3 Biotech* 9 (2):62. doi: 10.1007/s13205-019-1597-8.

Wang, F., H. Zhang, Z. Zhao, R. Wei, B. Yang, and Y. Wang. 2017. Recombinant lipase from *Gibberella zeae* exhibits broad substrate specificity: A comparative study on emulsified and monomolecular substrate. *International Journal of Molecular Sciences* 18 (7):1535. doi: 10.3390/ijms18071535.

Ward, O. P. 2012. Production of recombinant proteins by filamentous fungi. *Biotechnology Advances* 30 (5):1119–1139. doi: 10.1016/j.biotechadv.2011.09.012.

Weinhold, F. 2012. Natural bond orbital analysis: A critical overview of relationships to alternative bonding perspectives. *Journal of Computational Chemistry* 33 (30):2363–79. doi: 10.1002/jcc.23060.

Weissfloch, A. N. E., and R. J. Kazlauskas. 1995. Enantiopreference of lipase from *Pseudomonas cepacia* toward primary alcohols. *The Journal of Organic Chemistry* 60 (21):6959–69. doi: 10.1021/jo00126a056.

Whitaker, J. R. 1963. Determination of molecular weights of proteins by gel filtration of sephadex. *Analytical Chemistry* 35 (12):1950–3. doi: 10.1021/ac60205a048.

White, J. M., S. E. Delos, M. Brecher, and K. Schornberg. 2008. Structures and mechanisms of viral membrane fusion proteins: Multiple variations on a common theme. *Critical Reviews in Biochemistry and Molecular Biology* 43 (3):189–219. doi: 10.1080/10409230802058320.

Wilhelm, S., J. Tommassen, and K. Jaeger. 1999. A novel lipolytic enzyme located in the outer membrane of *Pseudomonas aeruginosa*. *Journal of Bacteriology* 181 (22):6977–86. doi: 10.1128/JB.181.22.6977-6986.1999.

Wilkes, R. A., and L. Aristilde. 2017. Degradation and metabolism of synthetic plastics and associated products by *Pseudomonas* sp.: Capabilities and challenges. *Journal of Applied Microbiology* 123 (3):582–93. doi: 10.1111/jam.13472.

Willems, N., M. Lelimousin, H. Koldsø, and M. S. P. Sansom. 2017. Interfacial activation of M37 lipase: A multi-scale simulation study. *Biochimica et Biophysica Acta. Biomembranes* 1859 (3):340–9. doi: 10.1016/j.bbamem.2016.12.012.

Williams, A. G., S. H. Beattie, and J. M. Banks. 2004. Enzymes involved in flavor formation by bacteria isolated from the smear population of surface-ripened cheese. *International Journal of Dairy Technology* 57 (1):7–13. doi: 10.1111/j.1471-0307.2004.00115.x.

Winkler, U. K., and M. Stuckmann. 1979. Glycogen, hyaluronate, and some other polysaccharides greatly enhance the formation of exolipase by *Serratia marcescens*. *Journal of Bacteriology* 138 (3):663–70. doi: 10.1128/jb.138.3.663-670.1979.

Wongwatanapaiboon, J., W. Malilas, C. Ruangchainikom, G. Thummadetsak, S. Chulalaksananukul, A. Marty, and W. Chulalaksananukul. 2016. Overexpression of *Fusarium solani* lipase in *Pichia pastoris* and its application in lipid degradation. *Biotechnology & Biotechnological Equipment* 30 (5):885–93. doi: 10.1080/13102818.2016.1202779.

Woodcock, L. L., C. Wiles, G. M. Greenway, P. Watts, A. Wells, and S. Eyley. 2008. Enzymatic synthesis of a series of alkyl esters using Novozymes 435 in a packed-bed, miniaturized, continuous flow reactor. *Biocatalysis and Biotransformation* 26 (6):466–72. doi: 10.1080/10242420802456571.

Woods, D. E., and J. U. Que. 1987. Purification of *Pseudomonas aeruginosa* exoenzyme S. *Infection and Immunity* 55 (3):579–86. doi: 10.1128/iai.55.3.579-586.1987.

Wu, X., C. Yang, and J. Ge. 2017. Green synthesis of enzyme/metal-organic framework composites with high stability in protein denaturing solvents. *Bioresources and Bioprocessing* 4 (1):1–8. doi: 10.1186/s40643-017-0154-8.

Xavier Malcata, F., H. R. Reyes, H. S. Garcia, C. G. Hill, Jr, and A. H. Clyde. 1990. Immobilized lipase reactors for modification of fats and oils—a review. *Journal of the American Oil Chemists' Society* 67 (12):890–910. doi: 10.1007/BF02541845.

Xu, X. 2000. Production of specific-structured triacylglycerols by lipase-catalyzed reactions: A review. *European Journal of Lipid Science and Technology* 102 (4):287–303. doi: 10.1002/(SICI)1438-9312(200004)102:4<287::AID-EJLT287>3.0.CO;2-Q.

Xu, Z., and S. Y. Lee. 1999. Display of polyhistidine peptides on the *Escherichia coli* cell surface by using outer membrane protein C as an anchoring motif. *Applied and Environmental Microbiology* 65 (11):5142–7. doi: 10.1128/AEM.65.11.5142-5147.1999.

Xu, Y., D. Lewis, and C. P. Chou. 2008. Effect of folding factors in rescuing unstable heterologous lipase B to enhance its overexpression in the periplasm of *Escherichia coli*. *Applied Microbiology and Biotechnology* 79 (6):1035–44. doi: 10.1007/s00253-008-1514-2.

Yaacob, N., N. H. A. Kamarudin, A. T. C. Leow, A. B. Salleh, R. N. Z. R. Abd Rahman, and M. S. Mohamad Ali. 2019. Effects of lid 1 mutagenesis on lid displacement, catalytic performances and thermostability of cold-active pseudomonas AMS8 lipase in toluene . *Computational and Structural Biotechnology Journal* 17:215–28. doi: 10.1016/j.csbj.2019.01.005.

Yadav, G. D., and S. Devendran. 2012. Lipase catalyzed synthesis of cinnamyl acetate via transesterification in a non-aqueous medium. *Process Biochemistry* 47 (3):496–502. doi: 10.1016/j.procbio.2011.12.008.

Yan, J., S. Liu, J. Hu, X. Gui, G. Wang, and Y. Yan. 2011. Enzymatic enrichment of polyunsaturated fatty acids using novel lipase prepa-

rations modified by combination of immobilization and fish oil treatment. *Bioresource Technology* 102 (14):7154–8. doi: 10.1016/j.biortech.2011.04.065.

Yang, T., X. Xu, C. He, and L. Li. 2003. Lipase-catalyzed modification of lard to produce human milk fat substitutes. *Food Chemistry* 80 (4):473–81. doi.org/ (02)00:473–81. 1 doi: 10.1016/S0308-8146.

Yeo, S.-H., T. Nihira, and Y. Yamada. 1998. Screening and identification of a novel lipase from *Burkholderia* sp. YY62 which hydrolyzes t-butyl esters effectively. *The Journal of General and Applied Microbiology* 44 (2):147–52. doi: 10.2323/jgam.44.147.

Yoshida, K., K. Konishi, A. Magana-Mora, A. Rougny, Y. Yasutake, S. Muramatsu, S. Murata, T. Kumagai, S. Aburatani, S.-I. Sakasegawa, et al. 2019. Production of recombinant extracellular cholesterol esterase using consistently active promoters in *Burkholderia stabilis*. *Bioscience, Biotechnology, and Biochemistry* 83 (10):1974–84. doi: 10.1080/09168451.2019.1630256.

Yu, M., S. Wen, and T. Tan. 2010. Enhancing production of *Yarrowia lipolytica* lipase Lip2 in *Pichia pastoris*. *Engineering in Life Sciences* 10 (5):458–64. doi: 10.1002/elsc.200900102.

Zaitsev, S., Yu, A. A. Savina, and I. S. Zaitsev. 2019. Biochemical aspects of lipase immobilization at polysaccharides for biotechnology. *Advances in Colloid and Interface Science* 272 (2019):102016. doi: 10.1016/j.cis.2019.102016.

Zalacain, I., M. J. Zapelena, M. P. De Peña, I. Astiasarán, and J. Bello. 1997. Use of lipase from *Rhizomucor miehei* in dry fermented sausages elaboration: Microbial, chemical and sensory analysis. *Meat Science* 45 (1):99–105. doi: 10.1016/S0309-1740(96)00049-6.

Zanger, U. M., and M. Schwab. 2013. Cytochrome P450 enzymes in drug metabolism: Regulation of gene expression, enzyme activities, and impact of genetic variation. *Pharmacology & Therapeutics* 138 (1):103–41. doi: 10.1016/j.pharmthera.2012.12.007.

Zdarta, J., A. S. Meyer, T. Jesionowski, and M. Pinelo. 2018. A general overview of support materials for enzyme immobilization: characteristics, properties, practical utility. *Catalysts* 8 (2):92. doi: 10.3390/catal8020092.

Zehani, N., R. Kherrat, S. V. Dzyadevych, and N. Jaffrezic-Renault. 2015. A microconductometric biosensor based on lipase extracted from *Candida rugosa* for direct and rapid detection of organophosphate pesticides. *International Journal of Environmental Analytical Chemistry* 95 (5):466–79. doi: 10.1080/03067319.2015.1036864.

Zhang, Y., A. N. A. Aryee, and B. K. Simpson. 2020. Current role of in silico approaches for food enzymes. *Current Opinion in Food Science* 31:63–70. doi: 10.1016/j.cofs.2019.11.003.

Zhangde, L., X. Jianhe, and P. Jiang. 2007. Immobilization of *Serratia marcescens* lipase and catalytic resolution of trans-3-(4′-methoxyphenyl) glycidic acid methyl ester. *Chinese Journal of Catalysis* 28 (2):175–9. doi: 10.1016/S1872-2067(07)60016-3.

Zhang, P., W. Zhang, X. Zhou, P. Bai, J. M. Cregg, and Y. Zhang. 2010. Catabolite repression of Aox in *Pichia pastoris* is dependent on hexose transporter PpHxt1 and pexophagy. *Applied and Environmental Microbiology* 76 (18):6108–18. doi: 10.1128/AEM.00607-10.

Zhao, L.-L., J.-H. Xu, J. Zhao, J. Pan, and Z.-L. Wang. 2008. Biochemical properties and potential applications of an organic solvent-tolerant lipase isolated from *Serratia marcescens* ECU1010. *Process Biochemistry* 43 (6):626–33. doi: 10.1016/j.procbio.2008.01.023.

Zhou, Y., M. Fan, and L. Chen. 2016. Interface and bonding mechanisms of plant fiber composites: An overview. *Composites Part B: Engineering* 101:31–45. doi: 10.1016/j.compositesb.2016.06.055.

Zhou, Q., Z. Su, L. Jiao, Y. Wang, K. Yang, W. Li, and Y. Yan. 2019. High-level production of a thermostable mutant of *Yarrowia lipolytica* lipase 2 in *Pichia pastoris*. *International Journal of Molecular Sciences* 21 (1):279. doi: 10.3390/ijms21010279.

Zou, X., J. Huang, Q. Jin, Z. Guo, Y. Liu, L. Cheong, X. Xu, and X. Wang. 2013. Lipid composition analysis of milk fats from different mammalian species: Potential for use as human milk fat substitutes. *Journal of Agricultural and Food Chemistry* 61 (29):7070–80. doi: 10.1021/jf401452y.

Zoumpanioti, M., E. Merianou, T. Karandreas, H. Stamatis, and A. Xenakis. 2010. Esterification of phenolic acids catalyzed by lipases immobilized in organogels. *Biotechnology Letters* 32 (10):1457–62. doi: 10.1007/s10529-010-0305-x.

Zucca, P., R. Fernandez-Lafuente, and E. Sanjust. 2016. Agarose and its derivatives as supports for enzyme immobilization. *Molecules* 21 (11):1577. doi: 10.3390/molecules21111577.

Zucca, P., and E. Sanjust. 2014. Inorganic materials as supports for covalent enzyme immobilization: Methods and mechanisms. *Molecules (Basel, Switzerland)* 19 (9):14139–94. doi: 10.3390/molecules190914139.

Hatzisymeon, M., S. Kamenopoulos, and T. Tsoutsos. 2019. Risk assessment of the life-cycle of the used cooking oil-to-biodiesel supply chain. *Journal of Cleaner Production* 217:836–43. doi: 10.1016/j.jclepro.2019.01.088.