Enzyme and Microbial Technology 112 (2018) 14–21

Contents lists available at ScienceDirect

Enzyme and Microbial Technology

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

D-Xylose and L-arabinose laurate esters: Enzymatic synthesis, characterization and physico-chemical properties

Thomas Méline[a], Murielle Muzard[b], Magali Deleu[c], Harivony Rakotoarivonina[a], Richard Plantier-Royon[b], Caroline Rémond[a,*]

[a] FARE laboratory, Chaire AFERE, Université de Reims-Champagne-Ardenne, INRA, 51686 Reims Cedex, France
[b] Institut de Chimie Moléculaire de Reims, CNRS UMR 7312, Université de Reims Champagne-Ardenne, 51687 Reims Cedex, France
[c] Université de Liège, Gembloux Agro-Bio Tech, Laboratoire de Biophysique Moléculaire aux Interfaces, 2 Passage des Déportés, B-5030 Gembloux, Belgium

ARTICLE INFO

Keywords:
D-Xylose laurate esters
L-Arabinose laurate esters
Lipase
Surfactants
Biorefinery

ABSTRACT

Efficient enzymatic synthesis of D-xylose and L-arabinose lauryl mono- and diesters has been achieved by transesterification reactions catalysed by immobilized *Candida antarctica* lipase B as biocatalyst, in organic medium in the presence of D-xylose or L-arabinose and vinyllaurate at 50 °C. In case of L-arabinose, one monoester and one diester were obtained in a 57% overall yield. A more complex mixture was produced for D-xylose as two monoesters and two diesters were synthesized in a 74.9% global yield. The structures of all these pentose laurate esters was solved. Results demonstrated that the esterification first occurred regioselectively onto the primary hydroxyl groups. Pentose laurate esters exhibited interesting features such as low critical aggregation concentrations values all inferior to 25 µM. Our study demonstrates that the enzymatic production of L-arabinose and D-xylose-based esters represents an interesting approach for the production of green surfactants from lignocellulosic biomass-derived pentoses.

1. Introduction

For several years, there has been an increasing interest for bio-based surfactants derived from annually renewable resources [1]. Among bio-based surfactants, alkyl glycosides and sugar fatty esters are non-ionic surface active compounds which present numerous advantages such as no toxicity for humans and for the environment, biodegradability, absence of odor and color [2–4]. The main fields of application of these non-ionic surfactants are related to personal care, cosmetics and pharmaceutical applications as well as food emulsification in case of sugar fatty esters [4–6]. The main alkyl glycosides and sugar fatty esters industrially produced or described in literature were generally obtained from hexoses, especially D-glucose, or hexose-based oligosaccharides such as sucrose, maltose and maltodextrins [1,7].

D-Xylose and L-arabinose are both pentoses abundantly present in lignocellulosic plant cell walls and are main components of xylans [8]. The production of new added-value molecules from pentoses represents a challenge for the valorisation of lignocelluloses in the context of development of biorefineries [1,7]. Xylose is currently reduced into xylitol, converted into furfural or fermented into ethanol [7]. Although few pentose-based surfactants were previously developed, some recent studies described the chemical synthesis of alkyl pentosides and pentose-based fatty esters [9–13]. These syntheses often require high energy and various catalysts that might not be compatible with green chemistry processes.

Classical chemical routes to the formation of fatty acid esters generally require esterification or transesterification reactions and the use of polar solvents (DMF, DMSO), a basic catalyst and high reaction temperatures (80–120 °C) leading to complex mixtures of monoesters, di- and higher esters as different regioisomers and unreacted sugar [14]. The use of fatty esters or acyl chlorides in the presence of an organic solvent and pyridine can improve the reaction yields but again with various degree of substitution [15]. Selective protection of the hydroxyl groups of the carbohydrate (acetyl, benzyl, isopropylidene) can orient the position of esterification reaction and also sometimes control the pyranose/furanose structure of the products [16].

The use of enzymes represents an interesting alternative for the preparation of surfactants from biomass [7,17]. For example, we previously described the enzymatic synthesis and the surfactant properties of alkyl xylosides and alkyl oligoxylosides from xylans and pretreated wheat bran using a transglycosylation approach with a xylosidase (EC 3.2.1.37) or xylanases (EC 3.2.1.8) [18,19]. Among well-known biocatalysts, lipases (EC 3.1.1.3) were widely used to catalyze the ester bond formation of sugar fatty esters [20,21]. Enzymatic synthesis of

* Corresponding author.
E-mail address: caroline.remond@univ-reims.fr (C. Rémond).

https://doi.org/10.1016/j.enzmictec.2018.01.008
Received 25 July 2017; Received in revised form 15 January 2018; Accepted 16 January 2018

sugar fatty esters is usually achieved by esterification reaction from a carbohydrate and a fatty acid or by a transesterification reaction from a carbohydrate and a fatty acid ester. Enzymatic synthesis of sugar fatty esters with lipases represents a green alternative compared to the conventional chemical approach [20]. Moreover, lipases can be used in immobilized form, then allowing a recyclability and reusability of the biocatalyst thus reinforcing the green alternative. Lipases display high regioselectivity compared to chemical acylation decreasing the complexity of mixtures of regioisomers produced [22–24]. During esterification, the amount of water present in reaction mixtures and formed during reaction must be highly controlled as water induces hydrolysis of esters products [25]. Hence, most of the lipase-based syntheses were performed in organic media as water quantity can be controlled by the use of salts or molecular sieves as desiccating agents [26,27]. Transesterification catalysed by lipases in presence of fatty acid esters, especially vinyl esters, represents an interesting strategy to overcome the water production during reaction and to induce better reactions yields. The main disadvantage in this latter case is the production of acetaldehyde as a by-product but the most widely employed lipases seem to be quite stable in the presence of acetaldehyde [28].

Although enzymatic synthesis of hexose-derived fatty esters was extensively described in the last twenty years, studies dealing with pentose-based fatty esters were less reported in literature [22,29–34]. Moreover, in most of the cases, structural data related to these molecules are not described.

In the present paper, we report the successful enzymatic synthesis of laurate pentose esters from D-xylose and L-arabinose catalysed by the lipase B from *Candida antarctica* (Novozym 435). Our strategy was based on transesterification reactions with vinyllaurate and allowed producing different mono- and diesters from D-xylose and L-arabinose. The structural features of these sugar esters were analysed by NMR and mass spectrometry and their surface-active properties were evaluated.

2. Experimental section

2.1. Materials

2-Methylbutan-2-ol (2M2B, 99%), molecular sieves (4 Å, beads, 8–12 mesh), hexane (> 95%), tetrahydrofuran (THF), Novozym 435 (immobilized lipase acrylic resin from *Candida antarctica*, Lot #SLBP0766V), vinyllaurate (> 99%), orcinol, chloroform (99%) and D-xylose (> 99%) were purchased from Sigma-Aldrich Corp. (St. Louis, USA). L-arabinose, acetic acid (AcOH, > 99%), ethylacetate (EA, > 99.8%), methanol (> 99.9%), petroleum ether (PE, > 99.9%) and *n*-butanol (BuOH, > 99%) were purchased from Roth (Karlsruhe, Germany). Sulfuric acid (H_2SO_4, 95%) was purchased from VWR (Radnor, USA). Acetonitrile (> 99.9%) and propan-2-ol (> 99%) were purchased from Carlo Erba Reagents (Dasit Group S.p.A, Cornaredo, Italy).

2.2. Methods

2.2.1. Enzymatic synthesis of D-xylose and L-arabinose laurate esters

Enzymatic syntheses were carried out in screwed glass bottles with magnetic stirrer, 400 × rpm, in an oil bath at 50 °C. Reactants, D-xylose or L-arabinose (50 mM) and vinyllaurate (150 mM) were incubated overnight with 2M2B and molecular sieves (10% w/v). Reaction started when Novozym 435 was added to the medium at 1% w/v. Reactions were stopped by incubating samples at 100 °C for 10 min and reaction mixtures were centrifuged at 500 × *g* for 5 min in order to pellet molecular sieves and enzymes. Supernatant was used to monitor sugar fatty esters production by thin layer chromatography and HPLC. Molecular sieves and enzymes were washed twice with ultrapure water in order to collect residual pentoses (HPLC quantification).

Kinetic studies were achieved at 50 °C with 20 mL reaction mix and sampling occurred at 1, 2, 4, 8, 24 and 48 h of incubation, 1 mL of reaction mixture was taken each time. These reactions were performed in triplicates.

Higher volume syntheses (100 mL) occurred in similar conditions in order to produce sufficient quantities of products for purification and characterization. Reactions were stopped after 4 h of incubation at 50 °C.

Recycling of the lipase was assessed in presence of D-xylose or L-arabinose (50 mM), vinyllaurate (150 mM), molecular sieves (10% w/v) and Novozym 435 (1% w/v). Reaction was conducted during 4 h at 50 °C under magnetic stirring (400 × rpm). After 4 h, reaction was centrifugated (45 × *g*) and pellets containing the lipase and the molecular sieves were further incubated with fresh D-xylose or L-arabinose and vinyllaurate. A total of 6 cycles of 4 h were performed.

2.2.2. Purification, characterization and quantification of the transesterification products

The production of pentose fatty esters was investigated by TLC, using pre-coated TLC-sheets ALUGRAM® Xtra SIL G/UV_{254} (Macherey-Nagel Gmbh & Co., Düren, Germany) and BuOH: AcOH: water (2/1/1) as the mobile phase. Products were revealed using 0.2% (w/v) orcinol in H_2SO_4 (20% v/v in water) and heating at 250 °C.

After removal of enzymes and molecular sieves, 2M2B was evaporated using a rotary evaporator (Büchi Labortechnik AG, Flawil, Switzerland) and crude products with remaining reactants were collected. Two hexane washings were then performed to eliminate the remaining vinyllaurate and a white paste was collected for both D-xylose and L-arabinose-based esters. Vinyllaurate removal was qualitatively assessed by HPLC. Finally, the residual pentose was precipitated in THF (100 mL) leading to soluble fractions containing mono- and diesters collected for further purification. Residual pentose was finally solubilized in water (50 mL) and quantified by HPLC.

The purification of D-xylose or L-arabinose laurate esters was performed by silica gel chromatography (9385 Merck Kieselgel 60, 230–400 mesh, 40–63 µm). Diesters were eluted using PE/EA (7/3, v/v) and monoesters were eluted using pure EA. All the products were obtained as a white crystalline powder.

NMR spectra were recorded on Bruker spectrometers (500 or 600 MHz for ^{1}H, 125 or 150 MHz for ^{13}C). Chemical shifts are expressed in parts per million (ppm) using tetramethylsilane as an internal standard. NMR spectra are presented as supplementary data. Mass spectra (ESI–MS) and high resolution mass spectra (ESI-HRMS) were performed on Q-TOF Micro micromass positive ESI (CV = 30 V).

2.2.2.1. 5-O-lauryl-L-arabinofuranose 1. White solid, mp 130 °C. ^{1}H NMR (500 MHz, $CDCl_3$): α/β = undetermined ratio, δ 5.30–5.35 (m, H-1α, H-1β), 4.20-4.35 (m, 2H-5α, 2H-5β, H-4α or β), 4.05-4.15 (m, H-2α, H-2β, H-4α or β), 2.37 (t, *J* = 7 Hz, 2H), 1.60-1.66 (m, 2H), 1.25-1.35 (m, 16H), 0.90 (t, *J* = 7 Hz, 3H); ^{13}C NMR (125 MHz, $CDCl_3$): δ 174.8 and 174.4 (C=O α and β), 102.5 (C-1α), 95.9 (C-1β), 82.5 and 81.2 (C-4 α and β), 79.5 (C-3β), 77.6 (C-2α), 77.4 (C-3α), 76.1 (C-2β), 65.9 and 64.3 (C-5 α and β), 2D experiment (HMBC): correlations between C=O and H-5; ESI–MS: 355.2 $(M + Na)^+$; ESI-HRMS: *m/z* calcd for $C_{17}H_{32}O_6Na$ 355.2097, found 355.2089.

2.2.2.2. 3,5-Di-O-lauryl-L-arabinofuranose 2. White solid, mp 69 °C, ^{1}H NMR (500 MHz, $CDCl_3$): α/β = 1/1.56, δ 5.35–5.40 (m, H-1α, H-1β), 4.94 (t, *J* = 5 Hz, H-3β), 4.70 (dd, *J* = 5 Hz, *J* = 2 Hz, H-3α), 4.29-4.44 (m, H-5α, H-5β, H-4α), 4.23 (dd, *J* = 12 Hz, *J* = 5 Hz, H-5α), 4.13-4.19 (m, H-2α, H-2β), 4.07-4.11 (m, H-4β), 2.38 (m, 2H), 1.59–1.65 (m, 2H), 1.20-1.38 (m, 16H), 0.90 (t, *J* = 7 Hz, 3H); ^{13}C NMR (125 MHz, $CDCl_3$): δ 174.6, 174.4, 173.9 and 173.5 (2C=O α and β), 102.9 (C-1α), 96.9 (C-1β), 81.2 (C-2), 81.0 (C-3 α), 79.9 and 79.8 (C-4α and C-3β), 79.1 (C-4β), 76.3 (C-2α and β), 65.0 and 63.4 (C-5 α and β), 2D experiments (HMBC): correlations between C=O 174.6 and 174.4 and H-3, C=O 173.9 and 173.5 and H-5; ESI–MS: 537.5 $(M + Na)^+$; ESI-HRMS: *m/z* calcd for $C_{29}H_{54}O_7Na$ 537.3767, found 537.3773.

2.2.2.3. 5-O-lauryl-D-xylofuranose 3a. White solid, mp 95 °C (lit. 93–95 °C [35]), ^{1}H NMR (600 MHz, CD_3OD): α/β = 1/1, δ 5.35 (d, *J* = 4 Hz, H-1α), 5.10 (d, *J* = 1 Hz, H-1β), 4.40 (dd, *J* = 11 Hz, *J* = 4 Hz, H-5β), 4.29-4.35 (m, H-4α and β), 4.27 (d, *J* = 4 Hz, H-5α), 4.25 (d, *J* = 4 Hz, H-5β), 4.13–4.18 (m, H-3α, H-5α), 4.05 (dd, *J* = 4 Hz, *J* = 2 Hz, H-3β), 3.96-3.98 (m, H-2β), 3.94 (t, *J* = 4 Hz, H-2α), 2.34 (q, *J* = 7 Hz, 2H), 1.59–1.65 (m, 2H), 1.25–1.35 (m, 16H), 0.90 (t, *J* = 7 Hz, 3H); ^{13}C NMR (150 MHz, CD_3OD): δ 175.5 and 175.4 (C═O α and β), 104.3 (C-1β), 97.8 (C-1α), 82.2 (C-2β), 80.9 (C-4β), 78.2 (C-2α), 77.7 (C-4α), 74.8 and 74.6 (C-3α and β), 65.4 (C-5α), 64.7 (C-5β), 2D experiment (HMBC): correlations between C═O and H-5; ESI–MS: 355.3 $(M + Na)^+$; ESI-HRMS: *m/z* calcd for $C_{17}H_{32}O_6Na$ 355.2097, found 355.2092.

2.2.2.4. 4-O-lauryl-D-xylopyranose 3b. White solid, mp 96 °C, ^{1}H NMR (500 MHz, CD_3OD): α/β = 2/1, δ 5.23 (d, *J* = 3 Hz, H-1α), 4.85 (dd, *J* = 8 Hz, *J* = 3 Hz, H-4β), 4.79 (td, *J* = 8 Hz, *J* = 5.5 Hz, H-4α), 4.72 (d, *J* = 6 Hz, H-1β), 4.14 (dd, *J* = 12 Hz, *J* = 5 Hz, H-5β), 3.94 (t, *J* = 8.5 Hz, H-3α), 3.72-3.83 (m, H-3β, 2xH-5α), 3.60 (d, *J* = 8.5 Hz, H-2α), 3.47 (t, *J* = 7 Hz, H-2β), 3.38 (dd, *J* = 12 Hz, *J* = 8.5 Hz, H-5β), 2.34 (q, *J* = 6 Hz, 2H), 1.58-1.65 (m, 2H), 1.25-1.35 (m, 16H), 0.87 (t, *J* = 7 Hz, 3H); ^{13}C NMR (150 MHz, CD_3OD): δ 173.7 (C═O α and β), 96.8 (C-1β), 92.4 (C-1α), 73.7 (C-2β), 72.9 (C-3β), 72.6 (C-2α), 71.7 (C-3α), 71.2 (C-4α), 71·1 (C-2β), 62.0 (C-5β), 59.7 (C-5α), 2D experiment (HMBC): correlations between C═O and H-4; ESI–MS: 327.2 $(M + H)^+$, 355.2 $(M + Na)^+$, 385.1 $(M + K)^+$; ESI-HRMS: *m/z* calcd for $C_{17}H_{32}O_6Na$ 355.2097, found 355.2089.

2.2.2.5. 2,5-Di-O-lauryl-D-xylofuranose 4. White solid, mp 79 °C, ^{1}H NMR (600 MHz, $CDCl_3$): α/β = 1/4.5, δ5.65 (d, *J* = 4 Hz, H-1α), 5.27 (broad s, H-1β), 5.07 (d, *J* = 1 Hz, H-2β), 4.93 (t, *J* = 4 Hz, H-2α), 4.56 (td, *J* = 9 Hz, *J* = 4.5 Hz, H-5β), 4.48 (dd, *J* = 12 Hz, *J* = 5 Hz, H-5α), 4.42 (q, *J* = 5 Hz, H-4α), 4.39 (dd, *J* = 5 Hz, *J* = 4 Hz, H-3α), 4.24-4.29 (m, H-5β, H-4β), 4.17 (dd, *J* = 12 Hz, *J* = 5 Hz, H-5α), 4.14-4.16 (m, H-3β), 2.30-2.39 (m, 4H), 1.58-1.62 (m, 4H), 1.19-1.39 (m, 32H), 0.90 (t, *J* = 7 Hz, 6H); ^{13}C NMR (150 MHz, $CDCl_3$): δ 175.0 (C═O β), 174.3 (C═O α), 173.8 (C═O α), 172.8 (C═O β), 100.9 (C-1β), 95.3 (C-1α), 81.1 (C-2β), 80.7 (C-4β), 79.9 (C-2α), 76.7 (C-4α), 74.2 (C-3α), 73.6 (C-3β), 62.6 (C-5β), 62.3 (C-5α), 2D experiment (HMBC): correlations between C═O (175.0 and 174.3) and H-2, and between C═O (173.8 and 172.8) and H-5; ESI–MS: 537.5 $(M + Na)^+$; ESI-HRMS: *m/z* calcd for $C_{29}H_{54}O_7Na$ 537.3767, found 537.3759.

2.2.2.6. 3,5-Di-O-lauryl-D-xylofuranose 5. White solid, mp 77 °C, ^{1}H NMR (500 MHz, $CDCl_3$): α/β = 1/0.2, δ 5.47 (d, *J* = 4 Hz, H-1α), 5.28 (d, *J* = 1 Hz, H-1β), 5.17 (dd, *J* = 5 Hz, *J* = 3 Hz, H-3α), 5.11 (dd, *J* = 5 Hz, *J* = 2 Hz, H-3β), 4.54-4.60 (m, H-4α, H-4β), 4.25-4.33 (m, 2H-5β), 4.10-4.22 (m,2H-5α, H-2α, H-2β), 2.25-2.35 (m, 4H), 1.55-1.64 (m, 4H), 1.20-1.28 (m, 32H), 0.80 (t, *J* = 7 Hz, 6H). ^{13}C NMR (125 MHz, $CDCl_3$): δ 173.80 (C═O α), 173.7 (C═O β), 173.5 (C═O α and β), 103.1 (C-1β), 96.0 (C-1α), 80.6 (C-2β), 78.6 (C-3α), 78.3 (C-3β), 77.8 (C-4β), 75.7 (C-2α), 75.0 (C-4α), 63.1 (C-5β), 62.2 (C-5α), 2D experiment (HMBC): correlations between C═O (173.8 and 173.5) and H-3, and between C═O (173.7 and 173.5) and H-5; ESI–MS: 537.5 $(M + Na)^+$; ESI-HRMS: *m/z* calcd for $C_{29}H_{54}O_7Na$ 537.3767, found 537.3777.

The quantification of sugar fatty esters was performed by HPLC using a NUCLEOSHELL® RP 18 ec, 5 µm 250 × 4.6 mm (Macherey Nagel) column at 40 °C. Purified products were used as standards. Mono- and diesters were eluted at 0.6 mL min^{-1}, at 40 °C and with an acetonitrile (**A**)/water (**B**) mobile phase, 0–5 min 80% **A** and 20% **B**, 5–20 min 80–100% **A** and 20–30 min 100% **A**. The detection was performed with a dynamic light scattering detector (PL-ELSD 1000, Polymer Laboratories) at 40 °C under 350 kPa azote pressure.

Retention times were determined for each ester **1**: 6.7 min, **2**: 31.5 min, **3a-b** (not separated in these conditons): 6.2 min, **5**: 28.9 min and **4**: 30.4 min.

The quantification of residual sugars was also performed by HPLC using a NUCLEODUR® 100-5 NH_2-RP, 5 µm 250 × 4 mm (Macherey Nagel) column at 40 °C. Pure sugars were used as standards. Sugars were eluted at 1 mL min^{-1}, at 40 °C and with an acetonitrile /water mobile phase 75/25% in isocratic flow. The detection was performed with a dynamic light scattering detector (PL-ELSD 1000, Polymer Laboratories) at 40 °C under 350 kPa azote pressure. Retention time of D-xylose and L-arabinose were 4.5 and 4.2 min respectively.

2.2.3. Determination of D-xylose and L-arabinose laurate esters surface-active properties

Adsorption to an air-aqueous medium interface was analysed at 25.0 ± 0.2 °C with an automated Langmuir Balance system equipped with a Wilhelmy plate (KSV minitrough, KSV instruments Ltd., Helsinki, Finland, 7.5 x 20 cm^2). Purified sugar esters, or mixtures of esters (extracted from crude reactions of 4 h at 50 °C with no further purification of mono- and diesters) were solubilized in DMSO and injected (20 µL) into the subphase (Tris HCl 10 mM pH 7) to a range of final concentrations (C). Injections were done using a Hamilton syringe and two homemade devices allowing the injection of the product without disturbing the air-water interface. These devices were placed at two fixed positions on the trough to ensure a reproducible injection process. The subphase was stirred, during the whole experimentation, using two cylindrical micromagnetic rods (8 ± 1.5 mm^2) and two electronic stirrer heads located beneath the trough (model 300, Rank Brothers, Bottisham, U.K.). Stirring was performed at 100 × rpm with an auto-reverse movement. The increasing of the surface pressure was recorded right after the injection until its value reaches equilibrium (ΔΠeq). CAC was determined from the plot ΔΠeq = f (C) at the intersection between the linear regression of the ascendant and plateau parts.

2.2.4. pH stability investigation

The pH stability of purified molecules was investigated by performing incubation at room temperature of 5 mM aqueous solutions of purified products at pH varying from 4 to 9. Citrate phosphate buffer 50 mM was used to prepare pH solutions from 4 to 8 and borate buffer 50 mM was used to prepare pH 9 solution. Stability was monitored, during 72 h with regular sampling (1, 2, 4, 8, 24, 48 and 72 h), qualitatively by TLC and quantitatively by HPLC.

3. Results

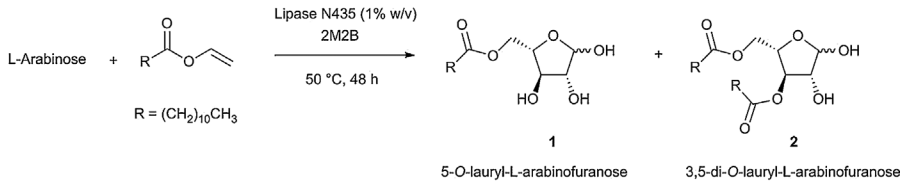
Transesterification reactions between L-arabinose or D-xylose and vinyllaurate were performed with immobilized *C. antarctica* lipase B, commercially known as Novozym 435. Various conditions were tested (substrates and lipase loading, organic solvents) and led to the choice of reaction parameters (data not presented) to carry out the enzymatic catalysis: L-arabinose or D-xylose (50 mM) and vinyllaurate (150 mM) with molecular sieves (10% w/v) in screwed glass bottles for various incubation times with 2M2B as solvent and magnetic stirring (400 × rpm).

3.1. Enzymatic synthesis of L-arabinose laurate esters

Products of transesterification reactions at 50 °C were first visualized by TLC, indicating that a mixture of monoester **1** and diester **2** of L-arabinose was obtained. Those esters were purified from 100 mL reactions performed during 4 h with conditions described above.

After purification by flash chromatography over silica gel and a careful NMR study, the structure of monoester **1** was attributed to 5-*O*-lauryl-L-arabinofuranose and the structure of diester **2** to 3,5-di-*O*-lauryl-L-arabinofuranose as mixtures of α/β anomers (Fig. 1A). Global conversion of L-arabinose was evaluated by HPLC by measuring L-arabinose remaining after 4 h in reaction media. From a 100 mL reaction at 50 °C, 320 and 756 mg of L-arabinose lauryl monoester **1** and diester **2**

A



B

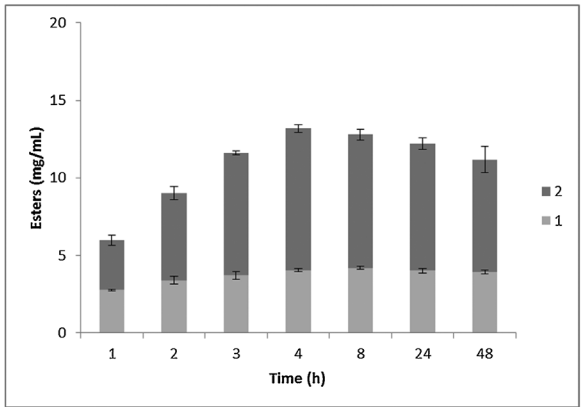


Fig. 1. (A) Transesterification reaction of L-arabinose and vinyllaurate performed by the lipase Novozym435. (B) Kinetics of synthesis of arabinose laurate esters quantified by HPLC. Reactions were performed at 50 °C during 48 h with 50 mM arabinose, 150 mM vinyllaurate, 1% (w/v) lipase. **1**: arabinose laurate monoester, **2**: arabinose laurate diester.

were respectively recovered corresponding to a conversion of 49.0% of introduced L-arabinose and a yield of 48.6% (19.2% of monoester **1** and 29.4% of diester **2**).

Kinetic of formation of L-arabinose lauryl esters obtained at 50 °C was evaluated for 48 h by HPLC with 20 mL reactions (Fig. 1B). During acylation of L-arabinose, both monoester **1** and diester **2** were formed during the first hour of the reaction (Fig. 2) and were produced at 2.8 ± 0.1 mg mL^{-1} and 3.2 ± 0.3 mg mL^{-1} respectively. Maximal quantities of L-arabinose lauryl esters were detected after 4 h of catalysis and were equivalent to 4.0 ± 0.1 and 9.1 ± 0.3 mg mL^{-1} for **1** and **2** respectively, corresponding to a conversion of 59.2 ± 1.5% of introduced L-arabinose and a 56.8% yield (22.8% of monoester **1** and 34.0% of diester **2**). After 4 h of reaction, a slight decrease in concentration of diester **2** was observed maybe due to a slight hydrolysis.

3.2. Enzymatic synthesis of D-xylose laurate esters

For the transesterification reactions of D-xylose with vinyllaurate at 50 °C during 4 h, two sets of spots were detected by TLC indicating the formation of a complex mixture of both monoesters and diesters. From 100 mL reactions, one major monoester **3a** was separated by flash chromatography and NMR data assigned its structure to the 5-*O*-lauryl-D-xylofuranose, as a mixture of anomers. A minor monoester **3b** was also detected and its structure was attributed to 4-*O*-lauryl-D-xylopyranose. Two diesters **4** and **5** were purified by flash chromatography over silica gel. The less polar diester **4** was identified as 2,5-di-*O*-lauryl-D-xylofuranose and the more polar diester **5** as 3,5-di-*O*-lauryl-D-xylofuranose as mixtures of anomers (Fig. 2A). From 100 mL reactions, a global conversion of 53% of initial D-xylose was reached. After separation by chromatography, 340 mg of monoesters (20.5%) and 557 mg of diesters **4** and **5** (25.5%) were purified and a global yield of 45.5% was obtained.

Kinetic studies were performed during 48 h in 20 mL reactions. During acylation reactions of D-xylose, the first products formed were monoesters **3a-b** 3.3 ± 0.3 mg mL^{-1} during the first two hours of the reaction (Fig. 2B). After the third hour, the two diesters appeared with quantities of 4.8 ± 0.1 mg mL^{-1} for **5** and 4.0 ± 0.1 mg mL^{-1} for **4** whereas the production of **3** reached 6.2 ± 0.1 mg mL^{-1}. The maximal production of xylose lauryl monoesters occurred in 4 h while the maximal yield of diester was attained after 24 h. Global conversion of D-xylose was evaluated by HPLC by measuring D-xylose remaining after 4 h in reaction media and attained 77.1 ± 2.0% and a 74.8% global overall yield (4 h) (38.4% of monoesters **3**, 19.4% of diester **4** and 17.4% of diester **5**). Conversion of D-xylose was higher compared to this obtained with the 100 mL reaction probably due to a better mass transfer.

Assays of recycling the enzyme were conducted in the same conditions, by performing transesterification of D-xylose and vinyllaurate during 4 h at 50 °C. Every 4 h, the reaction medium and the immobilized enzyme were separated. Reaction medium was analysed by HPLC and the remaining enzyme was used to perform another round of transesterification reactions with fresh D-xylose (50 mM) and vinyllaurate (150 mM). HPLC analysis showed that the Novozym 435 was reusable for 5 cycles without losing significant efficiency. During the sixth cycle, the synthesis of total D-xylose laurate esters reached 5% of synthesis occurring during each previous cycle indicating a drastic loss of lipase efficiency (data not presented). Similar results were obtained for recycling experiments in presence of L-arabinose.

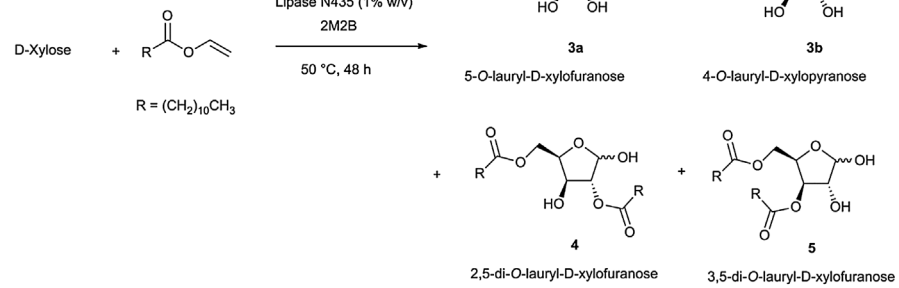
3.3. pH stability of L-arabinose and D-xylose laurate esters

The pH stability of the purified sugar esters produced at 50 °C was investigated by incubating aqueous solutions with pH range from pH 4–9. L-Arabinofuranose and D-xylofuranose laurate esters solutions were incubated at room temperature and samples were taken during 72 h. The evaluation of the stability was studied by HPLC (data not shown). The L-arabinofuranose and D-xylofuranose laurate esters were stable in solution within a pH range from 4 to 9 as no liberation of pentose indicating a hydrolysis of the ester bonds was detectable.

3.4. Surface-active properties of xylose and arabinose laurate esters

Surface-active properties were investigated for pentose esters produced at 50 °C. The ability of pentose esters to adsorb to an air-aqueous medium interface was studied by measuring the surface pressure increase further to the injection of the esters in the aqueous subphase. Fig. 3 shows as an example the adsorption kinetics of monoester **3a**. Similar results were obtained for the other esters. For concentration

A



B

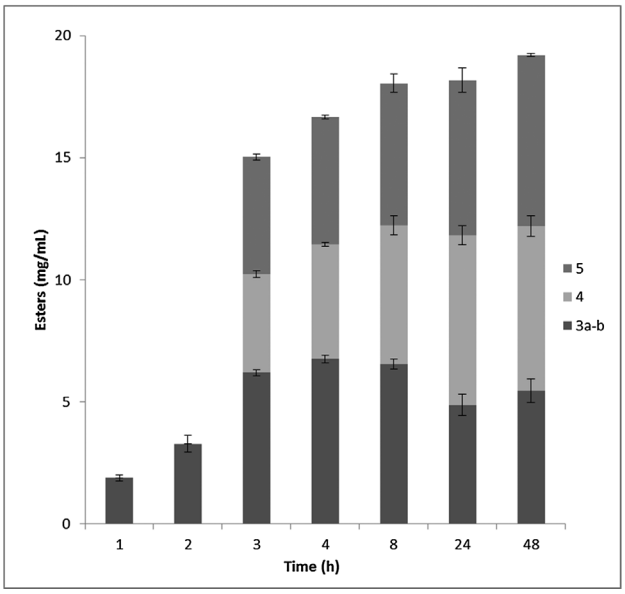


Fig. 2. (A) Transesterification reaction of D-xylose and vinyllaurate performed by the lipase Novozym435. (B) Kinetics of synthesis of xylose laurate esters quantified by HPLC. Reactions were performed at 50 °C during 48 h with 50 mM xylose, 150 mM vinyllaurate, 1% (w/v) lipase. **3a-b**: xylose laurate monoesters, **4** and **5**: xylose laurate diesters.

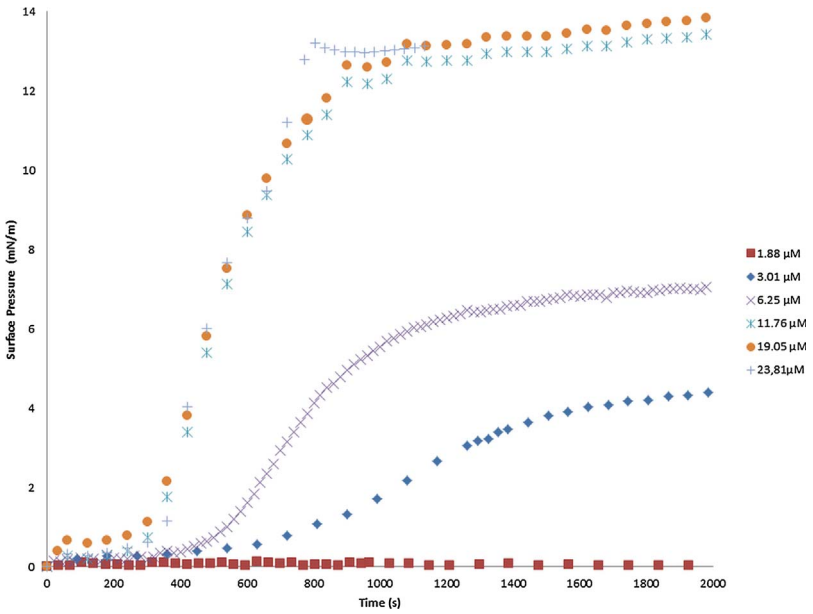


Fig. 3. Surface activity of the xylose laurate monoester **3a**. Kinetics of monoester adsorption at the air-water interface, time zero corresponds to the injection into the subphase.

Table 1
CAC values were measured for pure esters **1** and **3a** whereas in case of mixtures CAC values were determined with an average molecular weight of each compound.

	CAC
Xylose monoester (**3a**)	11.5 ± 1.6 µM
Arabinose monoester (**1**)	8.4 ± 0.9 µM
Xylose mono- and diesters mixture	23.4 ± 2.3 µM
Arabinose mono- and diester mixture	11.8 ± 0.7 µM

below 2 µM, no surface pressure increase was observed, suggesting that at very low concentration in the subphase, the adsorption of monoester **3a** to the air–water interface was too low to exert an effect on the surface pressure. At concentration ≥ at 3 µM, a surface pressure increase can be observed indicating that the monoester was able to adsorb at the air-aqueous medium interface. Before the detection of the surface pressure increase, there was a lag time, depending on the concentration (Fig. 3). This suggests that the adsorption to the air-aqueous medium interface is increased according to monoester **3a** concentration, indicating a higher probability of interactions between the ester molecules at the interface [36]. The higher number of molecules at the interface also explains the higher equilibrium surface pressure reached at the plateau.

Critical aggregation concentrations (CAC) were determined for purified D-xylofuranose and L-arabinofuranose monoesters and for mixtures of D-xylofuranose or L-arabinofuranose mono- and diesters (Table 1). For pure compounds, the surface pressure was measured for a range of concentrations going from 1 µM to 35 µM in the aqueous subphase (pH 7, room temperature) (Fig. 4). As pure D-xylofuranose diesters **4** and **5** or L-arabinofuranose diester **2** showed an extreme hydrophobic behavior (no solubility in aqueous media) for concentrations superior to 2 µM, no measurement of CAC was possible for these molecules. D-Xylofuranose monoester **3a** and L-arabinofuranose monoester **1** presented respectively a CAC of 11.5 ± 1.6 µM and 8.4 ± 0.9 µM (Fig. 4A and B). In case of xylose or arabinose esters mixtures, CAC were estimated to 23.4 ± 2.3 µM and 11.8 ± 0.7 µM respectively (Fig. 4C and D).

4. Discussion

Numerous studies from literature deal with esterification and transesterification reactions catalysed by lipases for hexoses acylation, mainly D-glucose. In this context, the selective acylation of the primary hydroxyl group for hexoses and the formation of 6-*O* acylhexopyranoses is well known [21,22,37].

The enzymatic transesterification of L-arabinose was previously described with a lipase from *Pseudomonas cepiaca* using oxime esters as acyl donors [38]. The reaction resulted in the selective formation of the 5-*O* monoacylated L-arabinofuranose in 45–70% yield according to the length of the acyl chain [38]. In a recent study, the synthesis of 5-*O*-palmitoyl-L-arabinofuranose monoester was obtained by esterification reaction with lipase N435 [34]. The regioselective acylation onto the most reactive primary hydroxyl position of the furanose isomer of L-arabinose was also obtained in the present study. However, in our reaction conditions, the diester **2** was also produced as the major compound in addition to the monoester **1**.

In case of D-xylose, lipase-catalysed esterification is described to yield complex mixtures of products with immobilized lipase of *C. antarctica* and lipase of *Candida rugosa* [32]. In previous studies, lipase-catalysed synthesis of fatty acid xylose esters was investigated but no information concerning neither the regioselectivity of the reaction nor the structures of the esters were available [32,33,39]. Lipase-catalysed esterification of 1,2-*O*-isopropylidene-α-D-xylofuranose with various fatty acids was studied. After deprotection of the acetal, selective synthesis of 5-*O*-acyl D-xylofuranose occurred [30,31,40]. Solvent-free esterification reactions at 75 °C with *Rhizomucor miehei* lipase and 1,2-*O*-isopropylidene-α-D-xylofuranose as acyl acceptor were carried out with fatty acids (lauric to arachidonic acid) and N435 as biocatalyst at 50 °C [40]. Conversion rate of the protected xylose reached 50% after 24 h with arachidonic acid.

In the present study, esters produced from D-xylose and L-arabinose at 50 °C were present as furanose isomers except a minor monoester **3b** for D-xylose as a pyranose isomer. No diesters in pyranosic form were detected and no acylation on the anomeric position occurred. This indicates that esterification first took place regioselectively onto the primary hydroxyl group. The pyranose/furanose equilibrium that should exist in the reaction mixture is displaced by the better reactivity of the primary hydroxyl group. Further esterification on a secondary hydroxyl group occurred with monoesters as substrates for the lipase. Indeed, experiments conducted with monoester **3a** as acyl acceptor at 50 °C (in the conditions described in section 2.1.1) showed that, after 2 h of reactions, diesters **4** and **5** were obtained in the same ratio (data not

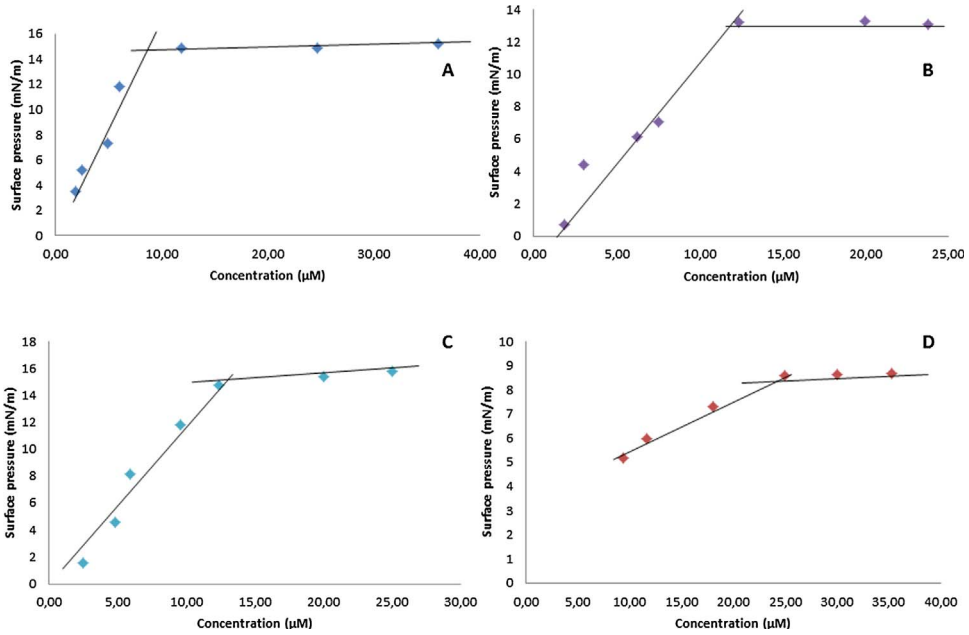


Fig. 4. CAC determination of purified arabinose and xylose laurate monoesters **1** and **3** (A and B respectively) and of mixtures of mono- and diesters of arabinose or xylose (C and D respectively). Results obtained at room temperature using a Langmuir balance equipped with a Wilhelmy platinum plate.

shown). For both pentoses, kinetic studies revealed that maximum yields were obtained at 50 °C after 4 h of reaction for xylose monoesters **3** and arabinose monoester **1** and diester **2** and after 24 h for xylose diesters **4**, **5**, with a higher conversion rate for D-xylose. However, D-xylose-based esters production was slower during the first two hours of reaction compared to L-arabinose. This could be explained by the lower ratio of the furanose isomers for D-xylose compared to L-arabinose in the reaction mixture as it is the case in aqueous solution [41] or by a difference in dissolution kinetics as described for D-glucose and D-fructose in 2M2B [42]. Monoesters **1** and **3a** can undergo a second acylation reaction. For L-arabinose, only diester **2** resulting from a transesterification reaction on the hydroxyl group at C-3 was synthesized and was already detected in the first hour of the reaction at 50 °C. No evolution of the monoester/diester ratio was observed during the reaction course even after 48 h. In comparison, the reaction was more complex for D-xylose, as two diesters **4** and **5** were obtained from monoester **3**. The second acylation proved to be less selective and slower than for L-arabinose. These results could be rationalized by an enhanced reactivity of the secondary hydroxyl groups on C-3 compared to the C-2 position for both monoesters **1** and **3a**. Indeed, for both monoesters, a second acylation was observed at C-3 position. However, for D-xylose monoester, the acylation on C-3 position could be in competition with acylation at C-2 maybe due to the *cis*-relationship(s) between OH in C-3 position and the acylated C-5 position which could generate steric hindrance.

The surface-active properties of the pentose-based esters produced at 50 °C can be compared to other sugar-based laurate esters described in literature. The CAC values were rather similar to those for xylofuranose laurate monoester for which CMC value of 41 μM or 18 μM have been determined [43,44]. Other laurate esters with different polar heads have shown variable CMC or CAC values depending on the polar head, such as maltopyranose and glucopyranose laurate ester with respective CMC values of 120 μM and 180 μM [44]. A CMC value of 3 mM was obtained for galactopyranose laurate esters [45]. Measurement of CMC/CAC is highly dependent on the pH and the temperature conditions. The CAC values obtained for our pentose-based laurate esters are inferior to 100 μM compared to hexose-based laurate esters and these low values could be related to the lower hydrophobicity of the smaller polar heads (pentoses *vs.* hexoses) [46].

The low CAC values of the D-xylofuranose and L-arabinofuranose esters produced in the present study indicate that these molecules present interesting surfactant properties as their CAC are lower than those of commercially available sugar esters such as Tween 20 (80 μM). Furthermore, the mixtures of esters also exhibit low CAC values which are of interest in a context of an industrial production with limited purification steps.

5. Conclusions

This study dedicated to the lipase-catalysed synthesis of fatty esters from two biomass-derived pentoses, D-xylose and L-arabinose, reports the full characterization of the esters produced and a better understanding of the regioselectivity of the lipase-catalysed transesterification with both pentoses. In this work, the enzymatic synthesis of D-xylose and L-arabinose laurate esters using *C. antarctica* lipase B (N435) as a biocatalyst was reported. Both L-arabinose and D-xylose gave rise to the synthesis of pentose laurate esters as furanose isomers indicating a regioselective first acylation onto the primary hydroxyl group. A second acylation could then occur either selectively on *O*-3 for L-arabinose or as a mixture of diacylated products on *O*-2 and *O*-3 positions for D-xylose. The nature of the pentose influenced kinetics of production and obtained yields. Kinetics of synthesis of D-xylose-based esters were slower but yields were higher than those obtained for L-arabinose-based esters.

Investigation of surfactant properties of pentoses laurate esters indicated that D-xylose and L-arabinose mono- and diesters exhibited good surfactant properties, such as low CAC, whether purified or in mixtures.

Our study allowed developing a green route for the one step functionalization of pentoses from lignocellulosic biomass to produce sugar esters that could be useful as surfactants for various applications (cosmetics, phytochemistry, food …).

Acknowledgements

The authors are grateful to the French Region Grand Est, Grand Reims and Feder for their financial support (chaire AFERE) as well as to the European Regional Development Fund (ERDF) and Region Grand Est for the financial support of the Interreg Valbran project. PhD thesis (T. Méline) was supported by the Region Grand Est.

M.D. thanks the F.R.S.-F.N.R.S. (National Funds for Scientific Research, Belgium) for her position as Senior Research Associate.

Appendix A. Supplementary data

Supplementary data associated with this article can be found, in the online version, at https://doi.org/10.1016/j.enzmictec.2018.01.008.

References

[1] P. Foley, E.S. Beach, J.B. Zimmerman, Derivation and synthesis of renewable surfactants, Chem. Soc. Rev. 41 (4) (2012) 1499–1518.
[2] I.J.A. Baker, B. Matthews, H. Suares, I. Krodkiewska, D.N. Furlong, F. Grieser, C.I. Drummond, Sugar fatty acid ester surfactants: structure and ultimate aerobic biodegradability, J. Surfactants Deterg. 3 (1) (2000) 1–11.
[3] S. De, S. Malik, A. Ghosh, R. Saha, B. Saha, A review on natural surfactants, RSC Adv. 5 (81) (2015) 65757–65767.
[4] W. von Rybinski, K. Hill, Alkyl Polyglycosides–properties and applications of a new class of surfactants, Angew. Chem. Int. Ed. 37 (10) (1998) 1328–1345.
[5] D.K.F. Santos, R.D. Rufino, J.M. Luna, V.A. Santos, L.A. Sarubbo, Biosurfactants multifunctional biomolecules of the 21st century, Int. J. Mol. Sci. 17 (3) (2016) 401.
[6] K. Hill, O. Rhode, Sugar-based surfactants for consumer products and technical applications, Lipid/ Fett. 101 (1) (1999) 25–33.
[7] R. Deutschmann, R.F.H. Dekker, From plant biomass to bio-based chemicals: latest developments in xylan research, Biotechnol. Adv. 30 (2012) 1627–1640.
[8] A. Ebringerova, T. Heinze, Xylan and xylan derivatives–biopolymers with valuable properties, 1. Naturally occurring xylans structures, isolation procedures and properties, Macromol. Rapid Commun. 21 (9) (2000) 542–556.
[9] F. Bouxin, S. Marinkovic, J. Le Bras, B. Estrine, Direct conversion of xylan into alkyl pentosides, Carbohydr. Res. 345 (17) (2010) 2469–2473.
[10] M. Deleu, C. Damez, S. Gatard, K. Nott, M. Paquot, S. Bouquillon, Synthesis and physico-chemical characterization of bolaamphiphiles derived from alkenyl d-xylosides, New J. Chem. 35 (10) (2011) 2258–2266.
[11] M. Deleu, S. Gatard, E. Payen, L. Lins, K. Nott, C. Flore, R. Thomas, M. Paquot, S. Bouquillon, D-Xylose-based bolaamphiphiles: synthesis and influence of the spacer nature on their interfacial and membrane properties, C. R. Chim. 15 (1) (2012) 68–74.
[12] N. Klai, C. Bidjou-Haiour, S. Bouquillon, D-Xylose-based surfactants: synthesis, characterization and molecular modeling studies, C. R. Chim. 18 (6) (2015) 599–606.
[13] F. Martel, B. Estrine, R. Plantier-Royon, N. Hoffmann, C. Portella, Development of agriculture left-overs: fine organic chemicals from wheat hemicellulose-Derived pentoses, Sustain. Dev. Top Curr. Chem. 294 (2010) 79–115.
[14] T. Polat, R.J. Linhardt, Syntheses and applications of sucrose-based esters, J. Surfactants Deterg. 4 (4) (2001) 415–421.
[15] O.T. Chortyk, J.G. Pomonis, A.W. Johnson, Syntheses and characterizations of insecticidal sucrose esters, J. Agric. Food Chem. 44 (6) (1996) 1551–1557.
[16] J. Guo, X.-S. Ye, Protecting groups in carbohydrate chemistry: influence on stereoselectivity of Glycosylations, Molecules 15 (10) (2010) 7235–7265.
[17] C. Dumon, L. Song, S. Bozonnet, R. Fauré, M.J. O'Donohue, Progress and future prospects for pentose-specific biocatalysts in biorefining, Process Biochem. 47 (3) (2012) 346–357.
[18] M. Muzard, N. Aubry, R. Plantier-Royon, M. O'Donohue, C. Rémond, Evaluation of the transglycosylation activities of a GH 39 β-d-xylosidase for the synthesis of xylose-based glycosides, J. Mol. Catal. B: Enzym. 58 (1–4) (2009) 1–5.
[19] M. Ochs, M. Muzard, R. Plantier-Royon, B. Estrine, C. Remond, Enzymatic synthesis of alkyl β-d-xylosides and oligoxylosides from xylans and from hydrothermally pretreated wheat bran, Green Chem. 13 (9) (2011) 2380–2388.
[20] S.W. Chang, J.F. Shaw, Biocatalysis for the production of carbohydrate esters, N. Biotechnol. 26 (3-4) (2009) 109–116.
[21] J.F. Kennedy, H. Kumar, P.S. Panesar, S.S. Marwaha, R. Goyal, A. Parmar, S. Kaur, Enzyme-catalyzed regioselective synthesis of sugar esters and related compounds, J. Chem. Technol. Biotechnol. 81 (6) (2006) 866–876.
[22] F. Cauglia, P. Canepa, The enzymatic synthesis of glucosylmyristate as a reaction model for general considerations on 'sugar esters' production, Bioresour. Technol. 99 (10) (2008) 4065–4072.
[23] P. Degn, L.H. Pedersen, J.O. Duus, W. Zimmermann, Lipase-catalysed synthesis of

glucose fatty acid esters in tert-butanol, Biotechnol. Lett. 21 (4) (1999) 275–280.
[24] I. Pérez-Victoria, J.C. Morales, Complementary regioselective esterification of non-reducing oligosaccharides catalyzed by different hydrolases, Tetrahedron 62 (5) (2006) 878–886.
[25] A.M. Klibanov, Improving enzymes by using them in organic solvents, Nature 409 (6817) (2001) 241–246.
[26] F. Chamouleau, D. Coulon, M. Girardin, M. Ghoul, Influence of water activity and water content on sugar esters lipase-catalyzed synthesis in organic media, J. Mol. Catal. B: Enzym. 11 (4-6) (2001) 949–954.
[27] G.D. Yadav, K.M. Devi, Immobilized lipase-catalysed esterification and transesterification reactions in non-aqueous media for the synthesis of tetrahydrofurfuryl butyrate: comparison and kinetic modeling, Chem. Eng. Sci. 59 (2) (2004) 373–383.
[28] H.K. Weber, H. Stecher, K. Faber, Sensitivity of microbial lipases to acetaldehyde formed by acyl-transfer reactions from vinyl esters, Biotechnol. Lett. 17 (8) (1995) 803–808.
[29] A.M. Gumel, M.S.M. Annuar, T. Heidelberg, Y. Chisti, Lipase mediated synthesis of sugar fatty acid esters, Process Biochem. 46 (2011) 2079–2090.
[30] G. Fregapane, D. Sarney, S. Greenberg, D. Knight, E. Vulfson, Enzymatic synthesis of monosaccharide fatty acid esters and their comparison with conventional products, J. Am. Oil Chem. Soc. 71 (1) (1994) 87–91.
[31] G. Fregapane, D.B. Sarney, E.N. Vulfson, Enzymic solvent-free synthesis of sugar acetal fatty acid esters, Enzyme Microb. Technol. 13 (10) (1991) 796–800.
[32] C. Tsitsimpikou, H. Daflos, F.N. Kolisis, Comparative studies on the sugar esters synthesis catalysed by *Candida antarctica* and *Candida rugosa* lipases in hexane, J. Mol. Catal. B: Enzym. 3 (1) (1997) 189–192.
[33] C. Bidjou-Haiour, N. Klai, Lipase catalyzed synthesis of fatty acid xylose esters and their surfactant properties, Asian J. Chem. 25 (8) (2013) 4347–4350.
[34] V.M. Pappalardo, C.G. Boeriu, F. Zaccheria, N. Ravasio, Synthesis and characterization of arabinose-palmitic acid esters by enzymatic esterification, Mol. Catal. 433 (2017) 383–390.
[35] J. Fernandez-Bolanos, F. Iglesias Guerra, C. Gomez Herrera, M. Lluch Colomer, Synthesis of special surfactants. VIII: synthesis of 3-O-acyl-D-xylopyranoses, 5-O-acyl-D-xylofuranoses and 3, 5-di-O-acyl-D-xylofuranoses, Tenside Deterg. 23 (3) (1986) 145–149.
[36] M.N. Nasir, A. Thawani, A. Kouzayha, F. Besson, Interactions of the natural antimicrobial mycosubtilin with phospholipid membrane models, Colloids Surf. B 78 (1) (2010) 17–23.
[37] A.M. Gumel, M.S.M. Annuar, T. Heidelberg, Y. Chisti, Thermo-kinetics of lipase-catalyzed synthesis of 6-O-glucosyldecanoate, Bioresour. Technol. 102 (19) (2011) 8727–8732.
[38] R. Pulido, F.L. Ortiz, V. Gotor, Enzymatic regioselective acylation of hexoses and pentoses using oxime esters, J. Chem. Soc. Perkin Trans. 1 (21) (1992) 2891–2898.
[39] E. Abdulmalek, N.F. Hamidon, M.B.A. Rahman, Optimization and characterization of lipase catalysed synthesis of xylose caproate ester in organic solvents, J. Mol. Catal. B: Enzym. 132 (2016) 1–4.
[40] O.P. Ward, J. Fang, Z. Li, Lipase-catalyzed synthesis of a sugar ester containing arachidonic acid, Enzyme Microb. Technol. 20 (1997) 52–56.
[41] K.N. Drew, J. Zajicek, G. Bondo, B. Bose, A.S. Serianni, 13 C-labeled aldopentoses: detection and quantitation of cyclic and acyclic forms by heteronuclear 1D and 2D NMR spectroscopy, Carbohydr. Res. 307 (3) (1998) 199–209.
[42] J.-M. Engasser, F. Chamouleau, L. Chebil, M. Ghoul, Kinetic modeling of glucose and fructose dissolution in 2-methyl 2-butanol, Biochem. Eng. J. 42 (2) (2008) 159–165.
[43] J. Fernandez Bolanos, F. Guerra, C. Herrera, M. Colomer, Synthesis of special surfactants, Chem. Inform. 18 (2) (1987).
[44] G. Garofalakis, B.S. Murray, D.B. Sarney, Surface activity and critical aggregation concentration of pure sugar esters with different sugar headgroups, J. Colloid Interface Sci. 229 (2) (2000) 391–398.
[45] D. An, X. Zhao, Z. Ye, Enzymatic synthesis and characterization of galactosyl monoesters, Carbohydr. Res. 414 (2015) 32–38.
[46] A. Ducret, A. Giroux, M. Trani, R. Lortie, Characterization of enzymatically prepared biosurfactants, J. Am. Oil Chem. Soc. 73 (1) (1996) 109–113.