Universiteit
Leiden
The Netherlands

# Transcriptional control of pectin degrading enzymes in Aspergillus niger

Niu, J.; Niu J.

**Citation**
Niu, J. (2017, October 18). *Transcriptional control of pectin degrading enzymes in Aspergillus niger*. Retrieved from https://hdl.handle.net/1887/54939

| | |
|---|---|
| Version: | Not Applicable (or Unknown) |
| License: | Licence agreement concerning inclusion of doctoral thesis in the Institutional Repository of the University of Leiden |
| Downloaded from: | https://hdl.handle.net/1887/54939 |

**Note:** To cite this publication please use the final published version (if applicable).

Cover Page

Universiteit Leiden

The handle http://hdl.handle.net/1887/54939 holds various files of this Leiden University dissertation.

**Author**: Niu, J.
**Title**: Transcriptional control of pectin degrading enzymes in Aspergillus niger
**Issue Date**: 2017-10-18

# Chapter 6

## An evolutionarily conserved transcriptional activator-repressor module controls expression of genes for D-galacturonic acid utilization in *Aspergillus niger*

Jing Niu, Ebru Alazi, Ian D. Reid, Mark Arentshorst, Peter J. Punt, Jaap Visser, Adrian Tsang, Arthur F.J. Ram

Published in Genetics. 2017 Jan; 205(1): 169-183

**Abstract**

The expression of genes encoding extracellular polymer-degrading enzymes and the metabolic pathways required for carbon utilization in fungi are tightly controlled. The control is mediated by transcription factors that are activated by the presence of specific inducers, which are often monomers or monomeric derivatives of the polymers. A D-galacturonic acid-specific transcription factor named GaaR was recently identified and shown to be an activator for the expression of genes involved in galacturonic acid utilization in *Botrytis cinerea* and in *Aspergillus niger*. Using a forward genetic screen, we isolated mutants that constitutively express GaaR-controlled genes. Reasoning that mutations in the *gaaR* gene would lead to a constitutively activated transcription factor, the *gaaR* gene in eleven of the constitutive mutants was sequenced, but no mutations in *gaaR* were found. Full genome sequencing of five constitutive mutants revealed allelic mutations in one particular gene encoding a previously uncharacterized protein (NRRL3_08194). The protein encoded by NRRL3_08194 shows homology to the repressor of the quinate utilization pathway identified previously in *Neurospora crassa (qa-1S)* and *Aspergillus nidulans* (QutR). Deletion of NRRL3_08194 in combination with RNA-seq analysis showed that the NRRL3_08194 deletion mutant constitutively expresses genes involved in galacturonic acid utilization. Interestingly, NRRL3_08194 is located next to *gaaR* (NRRL3_08195) in the genome. The homology to the quinate repressor, the chromosomal clustering, and the constitutive phenotype of the isolated mutants suggest that NRRL3_08194 is likely to encode a repressor, which we name GaaX. The GaaR-GaaX module and its chromosomal organization is conserved among ascomycetes filamentous fungi, resembling the quinate utilization activator-repressor module in amino-acid sequence and chromosomal organization.

**Key words:** gene regulation; galacturonic acid; repressor protein; genomics; transcriptomics; pectin

**Introduction**

The filamentous fungus *Aspergillus niger* is an important producer of pectin-degrading enzymes that are used in industrial applications including in food and feed processing (Kashyap *et al*. 2001; Khan *et al*. 2013). In nature, *A. niger* is a saprotrophic fungus that feeds on organic matter from decaying plants. The major carbon sources in plant cells are the storage polysaccharides starch, and less frequently inulin, as well as the cell wall polymers cellulose, hemicelluloses and pectin. Of the different plant polysaccharides, pectin has the most complex structure. Pectin is made up of four substructures including homogalacturonan, xylogalacturonan, rhamnogalacturonan I and rhamnogalacturonan II. The abundance of each substructure varies with plant species, but typically homogalacturonan is the most abundant polysaccharide in pectin (65%) followed by rhamnogalacturonan I (25-30%). Xylogalacturonan and rhamnogalacturonan II comprise less than 10% of the total pectin (Mohnen 2008).

Utilization of plant polysaccharides by fungi, including *A. niger*, is accomplished by tightly controlled secretion of extracellular enzymes that degrade the polymers into monosaccharides or oligosaccharides that are taken up and catabolised by the fungus. The controlled regulation is not only confined to the expression of genes encoding extracellular proteins. It also includes the controlled expression of genes encoding specific sugar transporters to guarantee efficient uptake of the liberated sugars and the intracellular catabolic pathway enzymes. The precise induction of the network of genes encoding substrate-specific enzymes, transporters and catabolic pathway enzymes has so far been shown to be mediated via $Zn(II)_2Cys_6$ transcription factors. Specific transcription factors in *A. niger* regulating the utilization of the major polysaccharides have been characterized. They include AmyR, the regulator for starch utilization (Petersen *et al*. 1999; Yuan *et al*. 2008a, vanKuyk *et al.* 2012); InuR for inulin (Yuan *et al*. 2008b); ManR, ClrA and ClrB for cellulose (Raulo *et al*. 2016); XlnR for xylan (Van Peij *et al*. 1998; Battaglia *et al*. 2014); AraR for arabinan (Battaglia *et al*. 2014); RhaR for rhamnose (Gruben *et al*. 2014); and GaaR for polygalacturonic acid (PGA) (Alazi *et al*. 2016). These transcription factors exert coordinated regulation of the target genes by interacting with conserved binding sites that are located upstream of the target genes. Computational analysis has been used to identify the galacturonic acid responsive element (GARE) of GA-induced genes (Martens-Uzunova and Schaap 2008). The predicted sequence (CCNCCAA) was shown to be required for the induction of GA-responsive genes in *A. niger* (Niu *et al*. 2015) and *Botrytis cinerea* (Zhang *et al*. 2016). Furthermore, using the

yeast one-hybrid method, it was shown in *B. cinerea* that the GaaR transcription factor interacts specifically with the GARE (Zhang *et al*. 2016).

Phenotypic characterization of mutants lacking the GA-regulator in both *B. cinerea* and *A. niger* has shown that GaaR is required for growth on GA (Zhang *et al*. 2016; Alazi *et al*. 2016). Expression analysis in both fungi confirmed that GaaR is required for the induced expression of GA-responsive genes. On complex pectins, growth of *B. cinerea* and *A. niger gaaR* deletion mutants was severely reduced and genome-wide expression analysis in *A. niger* revealed that the residual growth on pectin is likely due to the GaaR-independent expression of pectinases acting on arabinofuranosyl- and galactopyranosyl-containing side chains in rhamnogalacturonan (Alazi *et al*. 2016).

In addition to the transcription regulation via GaaR, GA-responsive genes are also under carbon catabolite repression (CCR) control (de Vries *et al*. 1999; de Vries *et al.* 2002). In filamentous fungi, CreA mediates CCR (Dowzer *et al*. 1991; Ruijter and Visser*,* 1997). In *A. niger*, CreA also exerts CCR control on GA-responsive genes (de Vries *et al*. 1999; Niu *et al*. 2015). Using an *'in vivo'* reporter construct consisting of the promoter of the GA-inducible *pgaX* gene (P*pgaX*) and the acetamidase (*amdS*) gene as a reporter, both the specific induction of *pgaX* on GA as well as the carbon repression of *pgaX* via CreA had been demonstrated (Niu *et al*. 2015). In this study, we have used the P*pgaX-amdS* reporter strain to isolate mutants displaying constitutive expression of GA-responsive genes. Analysis of the mutants resulted in the identification of a protein that likely acts as a repressor that specifically inhibits GaaR transcription activation activity under non-inducing conditions.

## Materials and Methods

### Strains, media and growth conditions

All strains in this study are listed in Table 1. Strains were grown in liquid or solidified (1.5% agar) minimal medium (MM) containing 7 mM KCl, 8 mM $KH_2PO_4$, 70 mM $NaNO_3$, 2 mM MgSO4 (pH adjusted to pH 5.5) as described (Bennett and Lasure, 1991). MM was supplemented with 50 mM glucose, 50 mM D-galacturonic acid, 50 mM fructose or 50 mM sorbitol as carbon source. Complete medium (CM) was also used and consists of MM supplemented with 0.1% casamino acids and 0.5% $w.v^{-1}$ yeast extract and 50 mM glucose. MM agar plates containing acetamide as sole nitrogen source were made as described previously (Arentshorst *et al*. 2012)

Table 1. *Aspergillus niger* strains used in this study.

| Name | Genotype/description | Reference/source |
|---|---|---|
| N402 | *cspA1*, derivative of N400 | Bos *et al.* 1988 |
| AB4.1 | *pyrG*$^-$ ,derivative of N402 | van Hartingsveldt *et al.* 1987 |
| MA234.1 | *Δku70::DR_amdS_DR* in MA169.4 | Alazi *et al.* 2016 |
| MA70.15 | *Δku70::amdS* in AB4.1 | Meyer et al., 2007 |
| MA299.2 | *Δku70* in MA70.15 | Niu *et al.* 2015 |
| MA323.1 | *Δku70::amdS, ΔnicB*$^-$*, pyrG*$^-$ | Niu *et al.* 2016 |
| JC1.5 | *pgaX-amdS* in MA299.2, *pyrG*$^+$ | Niu *et al.* 2015 |
| JN29.2 | *ΔcreA*::*hygB* in JC1.5 | Niu *et al.* 2015 |
| JN38 | spontaneous mutation S1in JN29.2 | This study |
| JN39 | spontaneous mutation S2 in JN29.2 | This study |
| JN42 | spontaneous mutation S5 in JN29.2 | This study |
| JN44 | spontaneous mutation S7 in JN29.2 | This study |
| JN52 | UV1 in JN29.2 | This study |
| JN53 | UV2 in JN29.2 | This study |
| JN54 | UV3 in JN29.2 | This study |
| JN55 | UV4 in JN29.2 | This study |
| JN56 | UV5 in JN29.2 | This study |
| JN57 | UV6 in JN29.2 | This study |
| JN58 | UV7 in JN29.2 | This study |
| JN59 | UV8 in JN29.2 | This study |
| JN60 | UV9 in JN29.2 | This study |
| JN61 | UV10 in JN29.2 | This study |
| JN62 | UV11 in JN29.2 | This study |
| JN63 | UV12 in JN29.2 | This study |
| JN64 | UV13 in JN29.2 | This study |
| JN122.1, JN122.1, JN122.1, | *ΔgaaX::phleo* in JN29.2 | This study |
| JN123.1, JN123.2, JN123.3 | *ΔgaaX::hygB* in JC1.5 | This study |
| JN125.1 | *ΔgaaX::nicB* in MA323.1 | This study |
| JN126.2, JN126.5, JN126.6 | PgaaX::GaaX::GFP::TgaaX in JN125.1 | This study |
| JN127.1, JN127.2, JN127.3 | PgaaX::GFP::GaaX::TgaaX in JN125.1 | This study |

**Isolation of mutants with constitutive expression of genes involved in polygalacturonic acid utilization**

*A. niger* strain JN29.2 (Table 1) was used for the selection of mutants with constitutive expression of genes involved in PGA utilization. Spontaneous mutants were obtained by plating out freshly harvested and myracloth filtered conidia (1 x $10^4$ conidia per plate) on MM glucose/acetamide plates and incubated at 30℃ for 5 days. In addition, mutants were obtained after mild UV mutagenesis (80% survival) as described (Damveld *et al*. 2008). Individual mutants growing on the primary MM-glucose/acetamide selection plates were purified twice on the MM-glucose/acetamide agar plates. In total, 14 spontaneous mutants and 59 UV-mutants were isolated that grew well on MM-glucose/acetamide agar plates and they were considered to be potential mutants with constitutive expression of genes involved in PGA utilization. To identify mutants constitutively producing PGA degrading enzymes, all 73 mutants were grown by inoculating 5 x $10^7$ spores in 50 ml MM-glucose medium for 36 h at

30℃ with shaking (150rpm). Supernatant of each culture was harvested by filtration. The extracellular culture fluid and the mycelia were stored at -80 ℃ for enzymatic assays and RNA extraction, respectively. Ten microlitres supernatant of each sample were spotted on PGA plates made by dissolving 0.2 % PGA (Sigma) in NaAc buffer (pH 4.2) with 1% agarose (Sphaero). The PGA plate assay was modified from the protocol used to detect cellulase activity (Teather and Wood 1982). Plates were incubated at 37℃ for 17 hours after spotting. PGA was stained by flooding the plates with a filter-sterile 0.05% solution of Congo Red (Sigma) dissolved in Milli-Q water for 15 min. The Congo Red solution was then poured off and the plates were washed with Milli-Q water, further treated by flooding with 1 M NaCl for 15 min. The formation of a clear zone of hydrolysis indicated PGA degradation.

The constitutive expression of genes involved in PGA degradation was further determined by northern blot analysis. Total RNA was isolated from eleven UV-mutants and two spontaneous mutants from frozen mycelia using TRIzol® reagent (Invitrogen). Quantification and purity assessment of total RNA was done by spectrophotometric method (NanoDrop 2000, Thermo Scientific). Total RNA, 3.5 µg, was loaded per sample and blotted to a Hybond$^{TM}$-N$^{+}$ nylon membrane (Amersham, GE Healthcare) followed by hybridization with [α-32P]-dCTP labelled probes (Rediprime II kit, Amersham, GE Healthcare). Probes were PCR-amplified using the N402 genomic DNA and the primer pairs are listed in Table S1. Standard molecular techniques were applied as described (Sambrook and Russell 2001).

**DNA sequencing and data analysis**

Sequencing of the *gaaR* gene from eleven constitutive mutants was performed by PCR amplification of the *gaaR* gene including 137 bp upstream and 152 bp downstream sequences using genomic DNA of the mutants as template and primers gaaRP7f and gaaRP8r (Table S1). Genomic DNA was isolated as described (Arentshorst *et al*. 2012). The PCR fragment (2765 bp in size) was sequenced in both directions using *gaaR* sequencing primers (Table S1). Sequencing was performed by Macrogen Europe (Amsterdam, The Netherlands).

Genomic DNA of three spontaneous mutants and two UV mutants was isolated as described (Arentshorst *et al*. 2012) and was further purified with DNA Isolation Kit (MO BIO Laboratories) for whole-genome DNA sequencing. The mutant genomes were sequenced at the McGill University Génome Québec Innovation Centre (Montreal) using the Illumina HiSeq platform to about 50-fold coverage. The DNA reads were aligned to the NRRL3

genome with Bowtie2 (Langmead and Salzberg 2012) and sequence differences were detected with Freebayes (Garrison and Marth 2012).

**Deletion of *gaaX* gene**

Deletion of the *gaaX* gene (NRRL3_08194) in the JC1.5, JN29.2 and MA323.1 backgrounds (Table 1) was carried out using the split marker approach (Arentshorst *et al*. 2015). The 869 bp 5'-flank and 870 bp 3'-flank regions were PCR amplified with the primers listed in Table S1 using N402 genomic DNA as template. These PCR fragments were used in fusion PCRs with hygromycin, phleomycin resistance genes or the *nicB* gene (Niu et al., 2016) to generate the split marker fragments. After amplification, the 5´flank-*hyg* and 3'flank-*hyg* fragments were transformed to the recipient strain JC1.5, the 5´flank-*phleo* and 3'flank-*phleo* fragments were transformed to the recipient strain JN29.2, and the 5´flank-*nicB* and 3'flank-*nicB* fragments were transformed to the recipient strain MA323.1. Putative *gaaX* disruption strains were purified by two consecutive single colony streaks. Genomic DNA was isolated as described (Arentshorst *et al*. 2012) and Southern blot hybridizations, using PCR-amplified fragments generated with primers listed in Table S1 as probes, were performed to confirm proper deletion and to exclude additional integrations.

**Bioreactor cultivation**

Controlled bioreactor cultivations for *A. niger* MA234.1 and JN123.1 were performed in 6.6-L BioFlo3000 bioreactors (New Brunswick Scientific) as previously described (Jørgensen *et al*. 2010). Briefly, autoclaved bioreactor vessels were filled with 5 L of sterile MM with 0.75% fructose. During cultivation at 30°C, the controller was set to maintain pH 3 by addition of titrants (2 M NaOH or 1 M HCl). Sterile air was supplied at a rate of 1 L $min^{-1}$. Prior to inoculation, 1.5 ml of 10% (w/v) filter-sterilized yeast extract was added to enhance conidial germination. Cultures were inoculated with freshly harvested spores at a concentration of 7,0 x $10^8$ conidia per liter. To reduce the loss of hydrophobic conidia during germination, the stirrer speed was set to 250 rpm and the culture was aerated via the headspace during the first six hours after inoculation. Subsequently, the stirrer speed was increased to 750 rpm, 0.5 ml of polypropyleneglycol P2000 was added as an antifoam agent and air was supplied via the sparger. Cultures broth was harvested at regular intervals from batch cultures and mycelial biomass was retained by vacuum filtration using glass microfiber filters (Whatman). Both biomass and filtrate were quickly frozen in liquid nitrogen and

subsequently stored at -80°C. Dry biomass concentrations were gravimetrically determined from lyophilized mycelia originating from a known mass of culture broth.

**Transcriptome analysis**

Mycelia grown in bioreactors to mid-exponential phase were used to isolate RNA using TRIzol® reagent (Invitrogen) and purified with NucleoSpin RNA Clean-up kit (Macherey-Nagel) with DNase treatment. Quantity and quality of the RNA samples were determined with a NanoDrop-2000 spectrophotometer and by RNA gel electrophoresis, respectively. RNA sequencing was conducted by Genome scan (Leiden, the Netherlands). Briefly, mRNA was isolated from the total RNA using NEBNext Ultra Directional RNA Library Prep Kit for Illumina according to the manufacturer's protocol. After fragmentation of the mRNA, cDNA was synthesized using random primers; and after a second strand cDNA synthesis reaction, fragments were ligated to the sequencing adapters. Clustering and DNA sequencing was performed using the Illumina NextSeq 500 SR75. Throughout the manuscript we will refer to *A. niger* Gene IDs based on the most up to date and accurate annotation of the *A. niger* NRRL3 genome (http://genome.fungalgenomics.ca/). The RNA-Seq reads were cleaned by correcting sequencing errors with Rcorrector (Song and Florea 2015), trimming sequencing adapters and low quality sequences with Skewer (Jiang *et al.* 2014), and removing ribosomal RNA with SortMeRNA (Kopylova *et al.* 2012). The cleaned reads were mapped to NRRL3 transcripts and counted with Salmon (Patro *et al.* 2016), and the read counts were analyzed for differences in transcript expression between genotypes with DESeq2 (Love *et al.* 2014).

**Construction of strains expressing GaaX-GFP or GFP-GaaX fusion proteins**

To construct fusions of GFP to the N-terminus or C-terminus of GaaX, PgaaX_GFP::GaaX_TgaaX and PgaaX_GaaX::GFP_TgaaX constructs were generated using a fusion-PCR approach in which N402 genomic DNA as well as plasmid PagsA_eGFP_TtrpC (Damveld *et al*. 2008) were used as template DNA. For constructing the PgaaX_GFP::GaaX_TgaaX construct, the promoter region of *gaaX* was PCR amplified using primers PgaaX_P7f-NotI and PgaaX_P11r, GFP was PCR amplified from plasmid PagsA_eGFP_TtrpC using primers GFP_P1f and GFP_P3r, *gaaX* and the terminator region of *gaaX* was PCR amplified using primers gaaX_P12f and TgaaX_P10r-NotI, and the three fragments were combined together in a two-step fusion PCR. Two amino acids (Gly-Ala) were introduced as spacer between GFP and GaaX. Subsequently, the fusion fragment was

cloned into vector pJet1.2 to give plasmid pJN34. For the PgaaX_GaaX::GFP_TgaaX construct, *gaaX* with the promoter region of *gaaX* was PCR amplified using primers PgaaX_P7f-NotI and gaaX_P8r, GFP was PCR amplified using primers GFP_P1f and GFP_P2r, the terminator region of *gaaX* was PCR amplified using primers gaaX_P9f and TgaaX_P10r-NotI, and the three fragments were combined together in a two-step fusion PCR. Again, a Gly-Ala spacer was introduced between GaaX and GFP. Subsequently, the fusion fragment was cloned into vector pJet1.2 to give plasmid pJN35.

Plasmids pJN34 and pJN35 were digested by *Not*I, and the fragments containing PgaaX_GFP::GaaX_TgaaX and PgaaX_GaaX::GFP_TgaaX were cloned into pMA334 (Arentshorst *et al*. 2015) to generate pJN36 and pJN37, respectively. The pMA334 plasmid has been designed such that the reporter constructs are targeted to the *pyrG* locus. Plasmids pJN36 and pJN37 were linearized by *Asc*I digestion and purified from gel before transformation to *A. niger* strain JN125.1. Proper integration of the PgaaX_GFP::GaaX_TgaaX or PgaaX::GaaX_GFP_TgaaX fragments at the *pyrG* locus was confirmed by Southern blot using PCR-amplified fragments generated with primers listed in Table S1 as probes.

**Microscopy**

For microscopic analysis, conidia of strains MA323.1, JN125.1, JN126.2 and JN127.3 were inoculated on coverslips in Petri dishes. Liquid minimal medium (MM) supplemented with 50 mM GA or 50 mM fructose as the carbon source was used. After incubation at 30℃ for 16 h, the coverslips with adherent germlings were mounted upside down on glass slides and observed under a confocal laser scanning microscope (Zeiss Imager, Zeiss, Jena, Germany), equipped with a LSM 5 exciter using 63x objectives. Images were processed by ImageJ with the exact same brightness and contrast adjustments and the median filter (radius 1.0).

**Data availability**

Strains are listed in Table 1 and are available upon request. Table S2 contains SNPs and indels detected in genomes of mutants. Table S3A contains TPM values of NRRL3 gene models in wild type and the *gaaX* mutant, and Table S3B contains their DEseq2 analysis. The DNA reads described in this study are deposited in the Short Read Archive under accession number SRP078415. The RNA reads described in this study are deposited in the Short Read

Archive under accession number SRP078485. The authors state that all data for confirming the conclusions presented in this article are represented fully within the article.

## Results

### Mutants constitutively expressing genes related to galacturonic acid utilization

To identify mutants that constitutively express genes related to PGA degradation and GA utilization in *A. niger*, we designed a forward screening procedure using a reporter strain containing a P*pgaX-amdS* reporter construct for positive selection of the desired mutants. We recently showed that the *pgaX* gene is specifically induced by GA, PGA and pectin, allowing the reporter P*pgaX-amdS* strain to grow on acetamide as a nitrogen source when GA, PGA or pectin is present as a carbon source (Niu *et al*. 2015). We also showed that deletion of the CCR protein (CreA) did not result in growth of the P*pgaX*-reporter strain on glucose, indicating that derepression via *creA* deletion was not sufficient to drive P*pgaX-amdS* expression to sustain growth. For the mutant screen, we used the P*pgaX-amdS* reporter strain in the Δ*creA* background (JN29.2) to prevent interference with possible CreA pathway-related repression mechanisms. Spores of *A. niger* strain JN29.2 were UV-mutagenized and surviving spores (80%) were plated on MM-glucose-acetamide plates. After mutagenesis, 59 mutants were isolated based on growth on acetamide. In addition to UV-generated mutants, 14 spontaneous mutants were isolated, resulting in a total of 73 mutants that could grow on glucose/acetamide plates.

To determine whether mutations were *cis*- or *trans*-acting, mutants were cultured in glucose medium for 36 h and the medium was analyzed for polygalacturonase activity. Initial experiments showed that cultivation of JN29.2 in glucose medium resulted in very low polygalacturonase levels and no halo was formed on Congo Red stained PGA plates when culture medium was spotted on a PGA plate (Figure 1A). We reasoned that if the mutation is *trans*-acting, the medium should contain increased levels of both exo- and endo-polygalacturonases leading to the formation of a halo. On the other hand, if the mutation is *cis*-acting, thereby only affecting the P*pgaX-amdS* reporter construct, it would not result in a halo on a PGA plate. Based on this assay, we concluded that the mutations of 65 out of the 73 mutants are *trans*-acting, while the remaining eight mutants carry presumed *cis*-acting mutations.

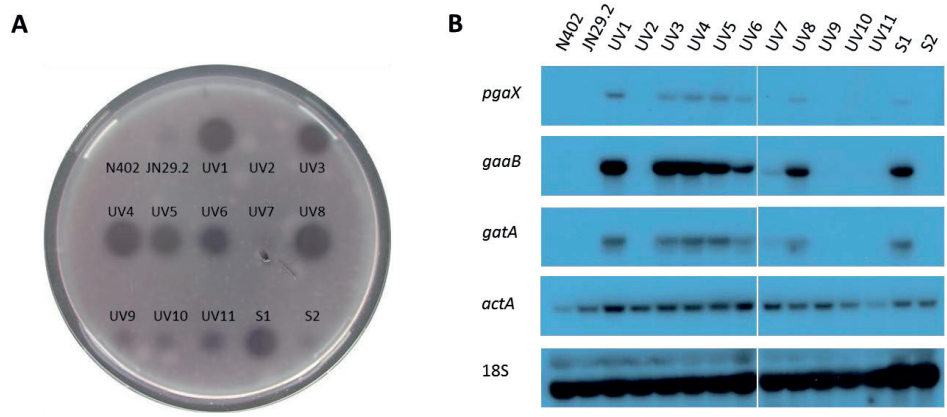


**Figure 1.** Enzymatic and RNA blot analysis of mutants with constitutive expression of genes involved in polygalacturonic acid utilization. A) 10 µl Supernatant from glucose-grown cultures of reference strains N402 and JN29.2, eleven UV mutants and two spontaneous mutants were spotted on polygalacturonic acid agarose medium to detect polygalacturonase activity. B) Northern blot analysis of selected GA-responsive genes in the reference strains N402 and JN29.2, eleven UV mutants and two spontaneous mutants.

To further demonstrate that the presumed *trans*-acting mutations indeed affected expression of multiple genes related to GA utilization and belonging to the GA-induced genes (Martens-Uzunova *et al*. 2008; Niu *et al*. 2015; Alazi *et al*. 2016), the expression of three GA-induced genes (*pgaX*, *gatA*, *gaaB)* was examined by northern blot analysis in a subset of mutants after growth on glucose. The *pgaX*, *gatA* and *gaaB* genes encode an exo-polygalacturonase, a GA-specific transporter, and the L-galactonic acid dehydratase involved in the GA release from PGA, the uptake of GA, and subsequent metabolism of GA, respectively. As shown in Figure 1B, expression of these genes was not detected in the wild-type (N402) and the parent JN29.2 (*ΔcreA*, P*pgaX-amdS*) strains whereas these genes were expressed in the constitutive mutants that displayed increased polygalacturonase activity. The presumed *cis*-acting mutants from the plate assay (UV2, UV7, UV9, UV10, UV11 and S2) did not constitutively express *pgaX*, *gatA* and *gaaB*, and showed a small halo on PGA plates, indicating the halo assay can be used to discriminate between *cis*- and *trans*-acting mutants. To determine whether a *cis*-acting mutation in the *pgaX* promoter in front of the *amdS* gene was responsible for the ability of this class of mutants to grow on acetamide, the *pgaX* promoter in front of the *amdS* gene of all eight cis-acting mutants was PCR amplified using *pyrG* and *amdS* specific primers (Table S1). This analysis revealed no mutations in the *pgaX* promoter region of any of the eight presumed *cis-acting* mutants. Hence, the nature of the

mutation(s) in these strains which allow growth on acetamide remains unknown. A possibility could be the activation of expression of endogenous *amdS* genes, as at least four *amdS*-like genes are present in the genome of *A. niger*.

**Identification of mutations responsible for the constitutive expression of the galacturonic acid utilization genes**

A possible explanation for the constitutive expression of GA utilization genes in the mutants is that they carry mutations in the recently identified GaaR transcriptional activator (Alazi *et al*. 2016). We therefore PCR amplified and sequenced the *gaaR* locus of eight constitutive mutants obtained after UV mutagenesis (UV1, UV3, UV4, UV5, UV6, UV8, UV12 and UV13) and three spontaneous mutants (S1, S5 and S7). The *gaaR* coding regions as well as 300 bp flanking regions were sequenced, but no mutation in the *gaaR* gene in any of these eleven mutants was found (data not shown).

To determine whether the constitutive expression of GA-induced genes in these mutants involves a functioning GaaR transcription factor, we deleted the *gaaR* gene in seven of the constitutive mutants (UV1, UV3, UV4, UV5, UV6, UV8 and S1) and analysed constitutive expression using the P*pgaX-amdS* reporter. All seven mutants were unable to grow on glucose/acetamide plates (data not shown), indicating that the constitutive expression of the GA-induced genes requires a functional *gaaR* gene.

To identify mutation(s) in the gene(s) responsible for the constitutive phenotype, the genomes of five mutants (UV1, UV8, S1, S5 and S7) and the parental strain JN29.2 were sequenced. Table 2 summarizes the number of SNPs and indels detected in the five mutant strains and Table S2 lists positions and type of all SNPs and indels detected. Spontaneous mutant S7 contains only eleven SNPs or indels, of which ten are located in intergenic regions and only one SNP mutated a gene, NRRL3_08194. Remarkably, in all four other mutants a mutation was found in the same gene. Two of the mutants carry nonsense mutations (UV1 and S5) and one a frame-shift mutation (S3), all leading to premature stop codons and predicted to result in truncated proteins (Table 2). Mutants S7 and UV8 have missense mutations in the C-terminal part of the protein. These results strongly suggest that the constitutive expression of genes encoding pectin-degrading enzymes in the five mutants is caused by a loss of function of the protein encoded by NRRL3_08194.

**Table 2.** Mutations in the constitutive mutants as compared to the parental strain JN29.2.

| Strain | Total number of SNPs and indels | SNPs or indels in coding region | Mutation in NRRL3_08194 | Position of mutation relative to ATG of NRRL3_08194 | Mutation in codon (bold) | Amino acid change | Predicted protein length (Full length protein is 697 amino acids) |
|---|---|---|---|---|---|---|---|
| JN29.2_UV1 | 40 | 19 | G-T | 1372 | **G**AG-**T**AG | E to Stop | 457 aa |
| JN29.2_UV8 | 21 | 4 | G-A | 1577 | G**G**A-G**A**A | G to E (526) | 697 aa |
| JN29.2_S1 | 68 | 34 | Extra G | 1958 | GTT-G**G**T | V to G out of frame | 663 aa |
| JN29.2_S5 | 48 | 11 | C-T | 1105 | **C**AA-**T**AA | Q to Stop | 368 aa |
| JN29.2_S7 | 11 | 1 | T-C | 2027 | C**T**G-C**C**G | L-P (676) | 697 aa |

**Deletion of NRRL3_08194 results in the constitutive expression of genes required for PGA breakdown and GA-catabolism**

To determine whether the constitutive expression of the target genes of the GaaR transcriptional activator (Alazi *et al*. 2016) is caused by a loss-of-function mutation in NRRL3_08194, we deleted this gene in the *pgaX-amdS* reporter strains JC5.1 and JN29.2 (*ΔcreA*), as well in a parental background without reporter constructs (MA323.1) (Table 1). The deletion mutants were purified and deletion of NRRL3_08194 was confirmed by Southern blot analysis for JC5.1, JN29.2 and MA323.1 (Figure S1 and S2). The verified deletion mutants were tested for growth on acetamide plates containing different carbon sources (Figure 2) as well as for constitutive expression of polygalacturonases (see below). Figure 2 shows that deletion of NRRL3_08194 in the *ΔcreA* background (JN122) resulted in the ability to grow on glucose/acetamide, fructose/acetamide and sorbitol/acetamide. The colony size on all three different carbon sources was similar, indicating that the *amdS* gene was expressed regardless of the carbon source used. However, deletion of NRRL3_08194 in the JC1.5 reporter strain (JN123) resulted in similar growth on fructose/acetamide and sorbitol/acetamide plates as JN122, but reduced growth on glucose. This indicates that glucose-mediated carbon catabolite repression repressed P*pgaX* driven *amdS* expression even in the absence of NRRL3_08194. The ability of the ΔNRRL3_08194 strain to grow on acetamide plates strongly suggests that a loss of function of NRRL3_08194 results in constitutive expression of *pgaX* and other pectinolytic genes. Furthermore, deletion of *gaaX* did not result in an altered growth behavior on GA, PGA, apple pectin, glucose, fructose, sorbitol, xylose and arabinose) (data not shown). These results are most easily explained by proposing that NRRL3_08194 encodes a repressor protein, which we name GaaX, that represses the activity of the GaaR transcription factor in the absence of GA. Interestingly, GaaX shows sequence similarity to a previously identified repressor protein, QutR (Grant et al., 1988). Moreover, the transcriptional activator (GaaR, NRRL3_08195) and the repressor (GaaX, NRRL3_08194) are clustered in the genome, similar to the quinic acid utilization transcriptional activator (QutA/Qa-1F) and repressor (QutR/Qa-1S) in *Aspergillus nidulans* and *Neurospora crassa,* respectively (Geever *et al*. 1989; Levesley *et al*. 1996).

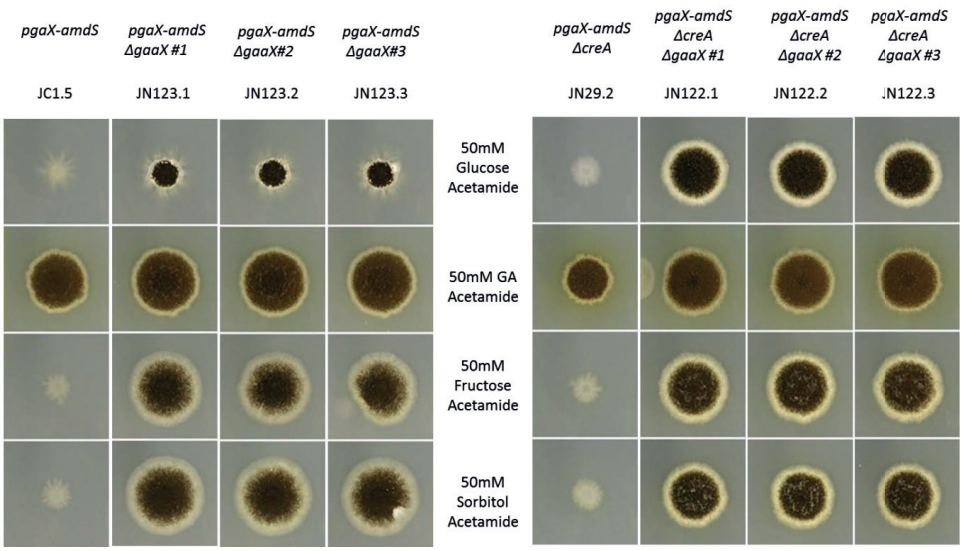


**Figure 2.** Regulation of the *pgaX* expression is controlled by GaaX and by CreA-mediated glucose repression. Growth of *pgaX-amdS* reporter strains in *gaaX* deletion and their parental strains was examined on various carbon sources. Parental strains and corresponding deletion strains were grown on MM-acetamide supplemented with 50 mM glucose, galacturonic acid (GA), fructose or sorbitol. All strains carry the P*pgaX-amdS* reporter construct. Strain JC1.5 is the parent of three independent transformants (JN123.1, JN123.2 and JN123.3) that contain a deletion in the *gaaX* gene. Strain JN29.2 carries the *ΔcreA* marker and is the parent of three independent transformants (JN122.1, JN122.2 and JN122.3) that contain a deletion in the *gaaX* gene.

## The GaaR-GaaX target gene regulon

We posit that the regulation of GA-responsive genes is likely to be negatively controlled by the repressor protein GaaX. A possible mode of action is that the repressor GaaX inhibits the activity of the transcriptional activator GaaR in the absence of an inducer. This would imply that deletion of the repressor or activation of the transcription factor by growth on GA would result in activation of the same set of genes. To show that the loss of function of the repressor activates the GA regulon and to identify the genes repressed by GaaX under non-inducing conditions, RNA-seq profiles of the *ΔgaaX* and its parental strain (MA234.1) were compared after growth on fructose, a non-repressing carbon source. Controlled cultivations in bioreactors showed that the growth rates ($\mu_{max}$ parental strain 0.214 ± 0.007 g dry weight $kg^{-1}$ $h^{-1}$ (n=3); $\mu_{max}$ *ΔgaaX* 0.223 ± 0.004 g dry weight $kg^{-1}$ $h^{-1}$ (n=2)) as well as biomass yields ($Y_{max}$ parental strain 4.15 ± 0.13 g dry weight $kg^{-1}$ (n=3); $Y_{max}$ *ΔgaaX* 4.29 ± 0.19 g dry weight $kg^{-1}$ (n=2)) of the two strains were highly comparable, indicating the *gaaX* deletion did not result in major physiological changes affecting the growth and biomass yield.

To identify differentially expressed genes in the *ΔgaaX* strain as compared to its parental strain, RNA-seq was performed on RNA isolated from exponentially growing cells at the time point at which about 75% to 80% of the maximum biomass yield was reached. RNA-seq reads were mapped to the NRRL3/N400 genome as this is the parent of the laboratory strain N402 and derivatives used in this study. Transcript per million (TPM) values were calculated using Salmon (Patro *et al,* 2016) (Table S3). Analysis of differential gene expression, based on a stringent False Discovery Rate (FDR)-value of <0.001 and a Fold Change (FC) >4.0, identified 37 upregulated genes (Table 3). Gene Ontology (GO) enrichment analysis using FetGOat (Nitsche *et al*. 2012) and manual inspection of the genes up-regulated in the *ΔgaaX* mutant indicated that genes involved in pectin catabolism were highly enriched. Of the 37 genes, sixteen are predicted to encode extracellular enzymes acting on the GA-backbone of pectin or acting on pectin side chains (Table 3). Nine genes in the group of 37 up-regulated genes in the *ΔgaaX* strain are predicted to encode intracellular proteins. Four of these nine genes (*gaaA*-*gaaD*) are required for the conversion of GA into pyruvate and glycerol (Martens-Uzunova and Schaap 2008). The exact role of the other five genes and their possible role in GA catabolism is currently unknown. The group of 37 up-regulated genes also includes seven genes predicted to encode sugar transporter proteins. Of these seven transporter-encoding genes, only GatA has been studied in detail and shown to be able to transport GA (Sloothaak *et al*. 2014). Apart from the genes encoding extracellular enzymes (16), transporters (7) and enzymes possibly involved in GA catabolism (9), the remaining five genes in this group encode proteins with unknown functions or with similarities to known proteins that for now cannot be directly linked to GA metabolism. The deletion of *gaaX* has the most profound effect on the transcript levels of the genes encoding the first three steps of the GA-utilization pathway (*gaaA*, *gaaB* and *gaaC*) and on the expression of *gatA*. Deletion of *gaaX* resulted in a 1.24-fold (P value 0.000035) increase in *gaaR* gene activity. Since the up-regulation of *gaaR* in the *ΔgaaX* mutant is modest, it is likely that the repressing activity of GaaX is mediated at the protein level (e.g. by interacting with GaaR) rather than by transcriptional control of *gaaR*. Seventeen of the 37 genes up-regulated in the *ΔgaaX* mutant were previously identified as part of the GA regulon (Martens-Uzunova and Schaap 2008; Alazi *et al*. 2016) (Table 3, Figure 3). Sixteen of the 17 genes found in common with previous studies are predicted or demonstrated to encode extracellular pectin-degrading enzymes. These results indicate that loss of function of *gaaX* affects the expression of the GA regulon The other 20 genes were identified as significantly up-regulated in the *gaaX* mutant, but these

were not identified previously as being part of the GA regulon (Table 3, Figure 3). A re-examination of the expression of these twenty genes in the RNA-seq data published earlier (Alazi *et al*. 2016) indicated that 10 of the genes (indicated in Table 3 by the asterisk, Table S4) were also GA induced or GaaR dependent for induction in this previous study. On the other hand, 15 genes identified to be GA-induced in a GaaR-dependent manner in the previous study (Alazi *et al*. 2016) were not significantly up-regulated in the *gaaX* deletion strain (Figure 3). These results therefore suggest that full induction of GA-inducible genes requires more than the loss of GaaX activity, and that an additional induction mechanism plays a role.

An additional GA-induced gene identified in the study of Martens-Uzunova and Schaap (2008) but missing in the GaaR study is *gaaX* itself. Expression of *gaaX* was not examined in the Alazi et al. (2016) study as its function was not yet directly linked to GA utilization. However, re-evaluation of the dataset revealed that the induced expression of *gaaX* on GA is dependent on GaaR (fold change of WT vs *ΔgaaR*: 18.7; P-value 0.003; Table S4). Combining the expression data of the *ΔgaaX* mutant (this study), the *ΔgaaR* mutant (Alazi *et al*. 2016) and the genes induced on GA (Martens-Uzunova and Schaap 2008), we propose a panregulon of 53 GaaR-GaaX controlled genes and a core GaaR-GaaX regulon of at least 27 genes (Figure 3, Table 3 and Table S4). These 27 genes include eleven genes present in the intersection of all three data sets, six genes present in the intersection of the *ΔgaaX* data and the *ΔgaaR* data (Alazi *et al*. 2016), nine genes identified by examining the *gaaX* dataset with supporting evidence from previous studies and *gaaX* (Figure 3). Of these 27 genes, all except NRRL3_00660 (carboxyesterase), NRRL3_10865 (alpha-N-arabinofuranosidase), NRRL3_03342 (short-chain dehydrogenase/reductase), NRRL3_08833 (hypothetical protein), and NRRL3_02479 (beta-galactosidase*)*, have at least one predicted GARE motif in the upstream regions of the coding region (Table 3). It is interesting to note that among the genes listed in Figure 3 and Table 3 that are up-regulated in the *ΔgaaX* some of them (NRRL3_00957 and NRRL3_00958; NRRL3_09862 and NRRL3_09863; NRRL3_03291 and NRRL3_03292) are clustered. Except for NRRL3_00958, which encodes a GA-specific transporter (Sloothaak et al. 2014), the possible role of these genes in pectin degradation is currently unknown.

**Table 3.** Comparative RNA-seq analysis between wild-type and *ΔgaaX* strains: genes (37) upregulated in *ΔgaaX* strain.

| Gene ID (NRRL3) | Gene ID (CBS513.88) | Gene name | Description | Average expression value wt[a] | Average expression value *ΔgaaX*[a] | FC *ΔgaaX* VS WT[b] | False Discovery Rate[b] | predicted localization | GARE element[c] |
|---|---|---|---|---|---|---|---|---|---|
| NRRL3_05649 | An02g07720[1,2] | *gaaC* | L-threo-3-deoxy-hexylosonate aldolase | 12.54 | 2283.77 | 169.39 | 0.00E+00 | intracellular | (-) -292, -606 |
| NRRL3_09863* | An11g03500 | | alpha-hydroxy acid dehydrogenase | 0.82 | 165.84 | 160.07 | 0.00E+00 | intracellular | (+)-543, (-)-182 |
| NRRL3_00958 | An14g04280[1,2] | *gatA* | MFS-type sugar/inositol transporter | 3.47 | 524.95 | 140.38 | 0.00E+00 | membrane | (+) -360 |
| NRRL3_06890 | An16g05390[1,2] | *gaaB* | L-galactonic acid dehydratase | 47.77 | 6256.70 | 121.49 | 0.00E+00 | intracellular | (+) -326 |
| NRRL3_03291 | An12g05600 | | heterokaryon incompatibility protein | 0.00 | 6.78 | 82.98 | 6.48E-78 | intracellular | (+)-737, -325 |
| NRRL3_05650 | An02g07710[1,2] | *gaaA* | D-galacturonic acid reductase | 19.92 | 1515.44 | 71.84 | 0.00E+00 | intracellular | (+) -414, -100 |
| NRRL3_03144 | An12g07500[1,2] | *pgaX* | exo-polygalacturonase | 1.36 | 51.63 | 32.66 | 2.60E-272 | extracellular | (+)-388 |
| NRRL3_05252 | An02g12505[2] | *pmeC* | pectin methylesterase | 1.06 | 31.32 | 25.03 | 7.10E-189 | extracellular | (+)-275, -246, -35 |
| NRRL3_06244* | An02g00140 | | glycoside hydrolase family 43 protein | 0.81 | 22.19 | 23.46 | 8.13E-193 | extracellular | (+)-96, (-)-712 |
| NRRL3_08281 | An03g06740[1,2] | *pgxB* | exo-polygalacturonase Pgx28B | 0.00 | 2.10 | 22.95 | 3.59E-43 | extracellular | (-) -298, -823 |
| NRRL3_10559 | An18g04810[2] | *rgxC* | glycoside hydrolase family 28 protein | 0.08 | 3.11 | 17.32 | 1.33E-45 | extracellular | (+)-852, (-) -250 |
| NRRL3_05260 | An02g12450[1,2] | *pgxC* | exo-polygalacturonase Pgx28C | 0.95 | 16.77 | 15.22 | 1.83E-144 | extracellular | (+) -268, (-) -642 |
| NRRL3_08663 | An03g01620[1,2] | | MFS-type sugar/inositol transporter | 0.27 | 5.62 | 14.28 | 8.28E-46 | membrane | (+) -673 |
| NRRL3_03342* | An12g04990 | | short-chain dehydrogenase/reductase | 0.81 | 14.46 | 13.51 | 2.10E-62 | intracellular | none |
| NRRL3_10865 | An08g01710[1,2] | *abfC* | alpha-N-arabinofuranosidase | 0.80 | 11.21 | 11.92 | 1.19E-84 | extracellular | none |
| NRRL3_09862 | An11g03510 | | hypothetical protein | 0.00 | 0.93 | 11.05 | 4.32E-18 | unknown | (-) -517, -829, -843 |
| NRRL3_10050 | An11g01120[1,2] | *gaaD/larA* | NADPH-dependent erythrose reductase Err1 | 256.41 | 2732.37 | 10.16 | 0.00E+00 | intracellular | (-) -538, -583, -801 -813 |
| NRRL3_00957 | An14g04260 | | B3/B4 domain-containing protein | 1.53 | 18.56 | 10.05 | 2.46E-64 | unknown | none |
| NRRL3_00502 | An09g06200 | | hypothetical protein | 0.93 | 12.38 | 9.36 | 1.08E-32 | unknown | (-) -189 |
| NRRL3_10558* | An18g04800 | | alpha-L-rhamnosidase | 0.35 | 3.85 | 9.09 | 2.19E-61 | extracellular | (+) -365 |
| NRRL3_11710 | An06g00620 | | MFS-type sugar/inositol transporter | 2.86 | 25.29 | 8.05 | 1.39E-118 | membrane | (+)-487, (-) -368 |
| NRRL3_06053 | An02g02540[1,2] | *paeA* | carbohydrate esterase family 16 protein | 2.06 | 17.62 | 7.76 | 5.39E-107 | extracellular | (+) -1238 |
| NRRL3_01073 | An14g05840 | | O-methyltransferase, COMT-type | 0.54 | 5.57 | 7.16 | 1.06E-21 | intracellular | (+) -300 |
| NRRL3_07382 | An16g00540 | | alpha-L-fucosidase | 0.04 | 0.67 | 7.07 | 5.92E-14 | extracellular | (-) -606 |

| | | | | | | | | | |
|---|---|---|---|---|---|---|---|---|---|
| NRRL3_00660* | An14g00860 | | carboxylesterase | 0.21 | 1.87 | 6.98 | 1.47E-31 | extracellular | None |
| NRRL3_08499 | An03g03960 | | uncharacterized protein | 0.48 | 4.58 | 6.71 | 3.84E-21 | unknown | none |
| NRRL3_07470 | An04g09690[2] | *pmeB* | pectin methylesterase | 0.75 | 5.72 | 6.36 | 3.59E-43 | extracellular | (+) -389 |
| NRRL3_02479 | An01g10350[2] | *lacB* | exo-beta-1,4-galactanase | 3.48 | 22.29 | 6.03 | 1.37E-179 | extracellular | none |
| NRRL3_03292 | An12g05590 | | carboxylesterase | 0.18 | 1.86 | 5.26 | 1.75E-09 | extracellular | (-) -1000 |
| NRRL3_01237 | An19g00270[2] | *pelD* | pectin lyase | 0.17 | 1.47 | 5.25 | 6.14E-14 | extracellular | (-) -409, -465 |
| NRRL3_03467 | An12g03550 | | MFS-type transporter | 1.13 | 6.73 | 5.05 | 4.03E-28 | membrane | none |
| NRRL3_01127* | An14g06500 | DHAK | dihydroxyacetone kinase | 18.90 | 100.64 | 4.98 | 1.61E-102 | intracellular | (+) -69 |
| NRRL3_11054* | An08g04040 | | MFS-type sugar/inositol transporter | 6.62 | 31.89 | 4.57 | 2.57E-126 | membrane | (+)-778, -726, (-) -646 |
| NRRL3_08833* | n.a. | | hypothetical protein | 0.31 | 2.02 | 4.52 | 1.58E-12 | unknown | none |
| NRRL3_01398 | An13g02090* | | MFS-type transporter | 2.81 | 13.10 | 4.16 | 5.62E-30 | membrane | (-) -270 |
| NRRL3_08325 | An03g06310[2] | *pmeA* | pectin methylesterase Pme8A | 0.04 | 0.56 | 4.10 | 1.49E-07 | extracellular | (+)-983, (-) -308 |
| NRRL3_02770 | An01g13880 | | MFS-type transporter | 0.87 | 4.07 | 4.06 | 1.57E-20 | membrane | (+) -578 |

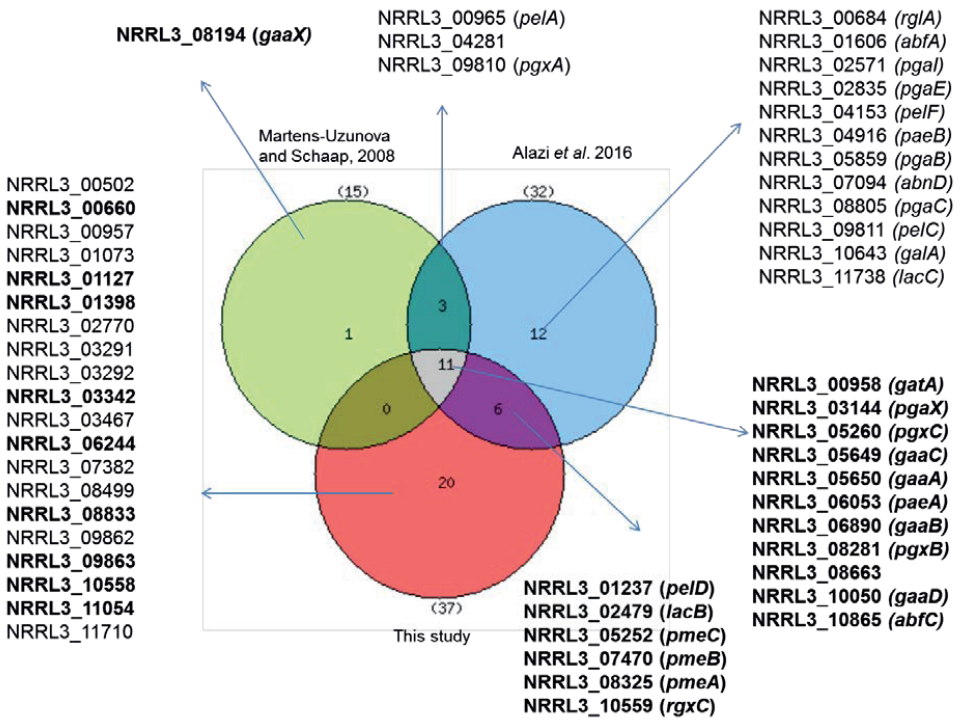


**Figure 3.** Venn diagram showing the overlaps between up-regulated genes in the wt_glucose versus wt_GA study (Martens-Uzunova and Schaap 2008), the up-regulated genes between *ΔgaaR*-GA versus wt-GA (Alazi *et al*. 2016), and the up-regulated genes in wt_fructose vs *ΔgaaX*-fructose (this study) to identify the GA-regulon. The 27 genes defining the GaaR-GaaX core regulon are indicated in bold.

### GaaX is induced on galacturonic acid and localized in the cytosol

GaaX was previously identified as a GA-induced gene with unknown function (Martens-Uzunova and Schaap 2008). To monitor the induction of GaaX and to localize the GaaX protein in the cell, GaaX was fused to GFP at either the N- or C-terminal part of GaaX and expressed from the endogenous GaaX promoter. Fusion constructs were targeted to the *pyrG* locus of *A. niger* in a strain lacking endogenous *gaaX* (JN125.1) to be able to test complementation of the GFP-GaaX and GaaX-GFP fusion proteins (Figure S3). As shown in Figure 4A, JN125.1 (*ΔgaaX::nicB*) constitutively expressed pectinases indicated by the halo on PGA plates, while both the C-terminally tagged as well as the N-terminally tagged versions of GaaX (JN126.2 and JN127.3 respectively) complemented the constitutive expression phenotype, indicating that both fusion proteins are functional. Confocal fluorescent microscopy was performed on GFP-tagged strains to localize GaaX (Figure 4B). Spores were germinated either on GA or on fructose (a non-repressing carbon source) and a

fluorescent signal was only detectable in the GFP-labeled strains after growth on GA. This observation confirms the results from the expression data that indicate that GaaX is lowly expressed under non-inducing conditions and is induced on GA. The expression of GaaX is low on fructose and no GFP signal above the background level was detected on fructose. Based on the fluorescent pictures, GaaX is likely to be localized in the cytosol.

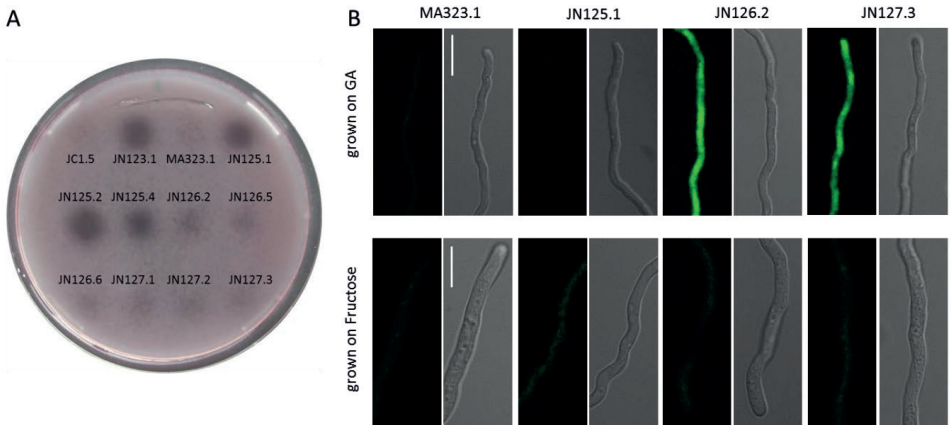


**Figure 4. A)** Complementation analysis of GaaX-GFP fusions. Polygalacturonase activities of *gaaX* deletion strains, *gaaX-GFP and GFP-gaaX* complementation strains, and their parental strains were detected by spotting 50 µl supernatant from fructose-grown cultures on polygalacturonic acid agarose. B) Subcellular localization of GaaX–GFP and GFP-GaaX in *A. niger* germlings. Strains were grown on coverslips in Petri dishes with minimal medium (pH 5.8) supplemented with either galacturonic acid or fructose as carbon source. Scale bar: 10µm.

## Discussion

The forward genetic screen with a positive selection strategy for the isolation of *A. niger* mutants with constitutive expression of genes involved in PGA degradation resulted in the identification of a repressor protein (NRRL3_08194) which we named GaaX. Both the genome sequencing of five independently obtained mutants as well as the analysis of a targeted deletion mutant (*ΔgaaX*), showed that the loss of function of *gaaX* leads to constitutive expression of genes previously identified as GA-induced genes (Martens-Uzunova and Schaap 2008) and genes encoding pectinolytic enzymes that are activated via the transcription factor GaaR (Alazi *et al*. 2016). Deletion of *gaaX* did not result in a growth alteration on any carbon source tested (Figure 2 and data not shown). Transcriptome analysis (Tables S3) strongly suggests that deletion of *gaaX* only affects the expression of genes related to the degradation and metabolism of (poly)galacturonic acid. Genes encoding

enzymes involved in the hydrolysis of non-pectin polysaccharides are not differentially regulated in Δ*gaaX*. In addition, GO enrichment analysis of Δ*gaaX* transcriptome shows a strong correlation only between the activity of GaaX and the expression of GA-induced genes. In agreement with these observations, the phenotype of the *gaaR* deletion mutant was specific for (poly)galacturonic acid with no growth defect observed on other substrates tested (glucuronic acid, rhamnose, xylose, arabinose) (Alazi *et al.* 2016). Taken together, these findings indicate the GaaR and GaaX are specifically involved in the regulation of pectin catabolism.

Interestingly, the *gaaX* gene is located next to the recently identified GA-specific transcriptional activator *gaaR* (NRRL3_08195). The GaaR transcriptional activator is conserved in 19 out of the 20 *Aspergillus* species for which genomic sequences are available via AspGD and only absent in *A. glaucus* (Alazi *et al*. 2016), which corresponds with the inability of *A. glaucus* to grow on GA (http://www.fung-growth.org/). In all nineteen *Aspergillus* species containing GaaR, a GaaX ortholog could be identified adjacent to GaaR. Only in *A. fumigatus* (Figure 5) and *A. wentii* (data not shown) were ORFs predicted to be present in between *gaaX* and *gaaR*. The ORFs between *gaaX* and *gaaR* in *A. fumigatus* are Afu4g06430 and Afu4g06450. Afu4g06430 is predicted to encode a 128 aa long protein which has no ortholog in other aspergilli. According to available expression data (Lind et al. 2015) this gene is not expressed. Whether this predicted gene actually encodes a protein is questionable. Afu4g06450 is predicted to encode a Tan1-related transposase of the DDE family. This type of transposase is found in both *A. nidulans* and *A. niger* as well as in many other organisms. This transposase is lowly expressed in *A. fumigatus* (Lind et al. 2015).

Like *gaaR*, *gaaX* is also missing in *A. glaucus*. BLASTP and synteny analysis between *A. niger* and *A. glaucus* revealed that the GaaR/GaaX encoding genes have been excised, as surrounding genes are conserved. Despite the loss of GaaX and GaaR, *A. glaucus* still possesses the GA-specific catabolic genes *gaaA* (Aspgl1_0124049), *gaaB* (Aspgl1_0091535) and *gaaC* (Aspgl1_0065497).

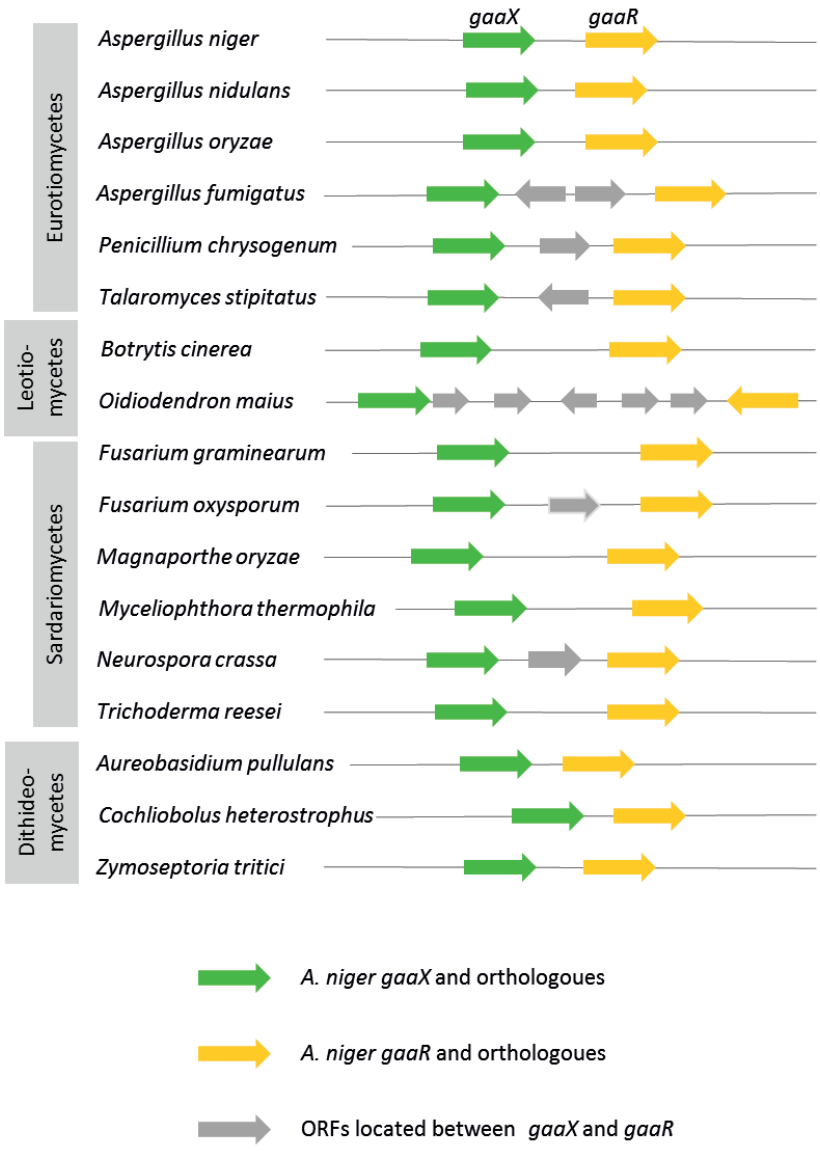


**Figure 5.** Schematic overview of the conservation of the *gaaX-gaaR* gene pair in 17 *Pezizomycotina* species. GaaX orthologs (green), GaaR orthologs (yellow) and ORFs between *gaaX* and *gaaR* (gray) are indicated. Arrow heads indicate the direction of transcription.

The GaaR transcriptional activator has previously been reported to be conserved in other Ascomycetes belonging to the Pezizomycotina subdivision, including members of the Eurotiomycetes (*Penicillium*, *Talaromyces* spp), Leotiomycetes (*Botrytis*, *Oidiodendron*), Sordariomycetes (*Neurospora*, *Myceliophthora*, *Magnaporthe*, *Trichoderma* and *Fusarium* spp.) and Dothideomycetes (*Zymoseptoria* (*Mycosphaerella*), *Aureobasidium* and *Cochliobolus* spp.) (Zhang *et al*. 2016). Synteny analysis of 17 species belonging to four classes of Pezizomycetes (Eurotiomycetes, Leotiomycetes, Sordariomycetes and Dothideomycetes) revealed a strong conservation of the genomic clustering of *gaaR* and *gaaX* orthologs (Figure 5 and Table S5). For most fungal species analysed, *gaaR* and *gaaX* are next to each other on the chromosome or close to each other and separated by one to five genes (Figure 5). The head to tail orientation of *gaaR-gaaX* driving expression of *gaaR* and *gaaX* from different promoters is conserved in all species except in *Oidiodendron maius*. Like GaaR, GaaX was found only in the Pezizomycotina and not in ascomycete yeasts, zygomycetes or basidiomycetes.

The strategy to identify the responsible mutation by sequencing five independently obtained mutants has been successful and efficient. Clearly, sequencing only a limited number of mutants leads only to successful identification when the mutants isolated in the screen all belong to a single complementation group. If more complementation groups are involved, more mutants would need to be sequenced. It is interesting to note that in addition to mutations in *gaaX* which were present in all five mutants, we noticed that two mutants (S1 and UV1) also contained allelic mutations in NRRL3_06175 (Table S2). The protein encoded by this gene is predicted to encode a cocaine esterase and belongs to a protein subfamily of hydrolases that included cocaine esterase (CocE), several glutaryl-7-ACA acylases, and the putative diester hydrolase NonD of *Streptomyces griseus.* This family shows extensive, low-level similarity to a family of Xaa-Pro dipeptidyl-peptidases. Whether this gene also contributes to the constitutive expression of GA-dependent genes remains to be determined, but this is unlikely as mutants without mutations in this gene display essentially the same constitutive phenotype.

Previous studies have identified genes specifically induced by GA (Martens-Uzunova and Schaap 2008) and pectinolytic genes that were dependent on the GaaR transcriptional activator for induction by GA (Alazi *et al*. 2016). Eleven of the fifteen GA-induced genes identified by Martens-Uzunova and Schaap were up-regulated in the *gaaX* mutant (Table 3 and Figure 3). The three genes that are considered GA-inducible but not detected as

differentially expressed in the *gaaX* mutant are predicted to encode a transporter (NRRL3_04281), an exo-polygalacturonase (NRRL3_09810, *pgxA*) and a pectin lyase (NRRL3_00965, *pelA*). These three genes were not classified as differentially expressed according to the stringent statistical settings in our current study. The fourth gene induced on GA in the study of Martens-Uzunova and Schaap (2008) but missing in our study is *gaaX* itself.

In our recent study on the GaaR transcriptional activator, we identified 32 pectinolytic genes whose expression on GA was dependent on GaaR (Alazi *et al*. 2016). These genes overlap largely with the previously identified GA-responsive genes (Martens-Uzunova and Schaap 2008) (Table 3, Figure 3), but also include eighteen new potential GaaR target genes. Six of these genes (including NRRL3_02479 (*lacB*), NRRL3_05252 (*pmeC*), NRRL3_08325 (*pmeA*), NRRL3_07470 (*pmeB)*, NRRL3_10559 (*rgxC*) and NRRL3_01237 (*pelD*) were also found to be significantly up-regulated in *ΔgaaX* (Table 3 and Figure 3) and are therefore considered to be part of the core GA-regulon. The remaining twelve genes identified as being GaaR dependent for induction on GA (Alazi et al. 2016) were not identified as differentially expressed based on the stringent settings in this study. Whether these genes are indeed directly controlled by GaaR and GaaX, and therefore part of the core GA regulon, awaits further study.

The GaaX protein is predicted to be 697 amino acids long and displays significant similarity to the last three domains in the C-terminal half of the AROM protein. AROM is a large (1586 amino acids in *A. niger*) pentafunctional protein composed of five domains and the individual domains are involved in five different enzymatic steps representing the prechorismate shikimate pathway, which is required for aromatic amino acid biosynthesis (Duncan *et al*. 1987; Hawkins and Smith 1991). The last three domains of the AROM protein encode the shikimate kinase (SK), 3-dehydroquinate dehydratase (DQ) and shikimate dehydrogenase (SDH) and are homologous to the respective bacterial enzymes (aroL, aroD and aroE) (Lamb *et al,* 1996). The AROM protein is present in fungi, including yeasts, and Euglena. The evolutionary origin of AROM is likely to be bacterial and it has been suggested that the AROM protein is the result of gene fusion events (Richards *et al*. 2006). Sequence alignment and BLASTP searches showed that the GaaX protein has significant sequence homology with the last three domains of the AROM protein. The observation of a transcriptional activator (GaaR) located next to a possible repressor protein (GaaX) that displays significant homology to AROM is analogous to the clustered transcriptional

activator/repressor module regulating quinic acid utilization (Geever *et al*. 1989; Lamb *et al*. 1990). Like GaaX, the quinate repressor protein shows significant sequence similarities with the last three C-terminal domains of AROM (Lamb *et al*. 1996).

The regulation of metabolic enzymes required for quinic utilization has been a classical example of gene regulation both in *N. crassa* and *A. nidulans* (Geever *et al*. 1989; Leversley *et al*. 1996). In *A. nidulans* and *N. crassa*, the transcriptional activator and repressor are located in a gene cluster which consists of the activator and repressor and other genes involved in quinic acid catabolism and transport (Geever *et al*. 1989; Lamb *et al*. 1990). *A. niger* also has a quinic acid gene cluster that includes, besides the *qutA* gene (NRRL3_11038) and *qutR* gene (NRRL3_11039), a catabolic 3-dehydroquinase (NRRL3_11037) and a MFS transporter possibly involved in quinate uptake (NRRL3_11036). In contrast to the quinic acid gene cluster in which the regulatory genes (activator and repressor) are clustered with structural genes, no structural genes involved in GA ultilization were clustered with GaaR and GaaX. Deletion of the *qutA* transcription factor (NRRL3_11038) in *A. niger* results in a quinate non-utilizing mutant (M. Arentshorst and A.F.J. Ram, unpublished results). Both in *A. nidulans* and *N. crassa*, the regulation of genes involved in quinic acid metabolism has been studied in detail and is characterized by the presence of a transcriptional activator (named QutA in *A. nidulans*, and *qa-1F* in *N. crassa*) located next to a repressor protein (QutR in *A. nidulans,* and *qa-1S* in *N. crassa*). Loss of function of quinic acid repressor *qutR* or *qa-1S* in *A. nidulans* and *N. crassa,* respectively, leads to constitutive expression of quinic acid utilization genes (Lamb *et al*. 1996; Giles *et al*. 1985), very similar to the effect observed for the loss of function of GaaX, resulting in constitutive expression of GA utilization genes. Based on the phenotype of the *gaaX* mutant and the analogy to the organization of the quinic acid utilization gene cluster, our current working hypothesis is that *gaaX* encodes a repressor protein which is required to keep the transcriptional activator GaaR in an inactive form in the absence of the inducer molecule.

As noted earlier, *gaaX* (NRRL3_08194) was identified as a up-regulated gene when an *A. niger* culture pregrown for 18 h with 2 % fructose was transferred to a medium containing 1% GA as the sole carbon source (Martens-Uzunova and Schaap 2008). The expression of a functional GFP-tagged version of GaaX confirmed the induced expression and showed cytosolic localization of GaaX in the presence of GA (Figure 4). In the promoter region of *gaaX*, a GA-responsive element (GARE) was found, suggesting that activation of the transcription factor results in increased levels of repressor protein. Although this might seem

contradictory at the first glance, it could actually be an elegant mechanism to ensure that the expression of GA-induced genes is tightly controlled and quickly responds to the presence or absence of GA. The induction of the expression of the repressor is partially analogous the activation/repression system of the qa cluster in *N. crassa*. In *N. crassa* it has been shown that both the activator (qa-1F) and the repressor (qa-1S) are transcriptionally induced in the presence of quinic acid (Patel et al. 1981; Giles et al. 1991). In the GA regulation system of *A. niger*, only the repressor protein is induced and not the activator. It should be noted that in almost all of 17 species analysed, the *gaaX* and *gaaR* genes do not share the same promoter region (head to tail orientation; Figure 5), while the *qa-1S* and *qa-1F* genes of *N. crassa* share the same promoter region, which might function as a bidirectional promoter.

As a working model (Figure 6), we postulate that in the presence of GA, the inducer molecule, which could be GA or a derivative of GA, binds in the cytosol to repressor protein GaaX. Binding of the inducer to the GaaX repressor is posited to result in the activation of the transcription factor GaaR. Active GaaR is expected to induce the expression of GA-responsive genes involved in GA release, uptake and metabolism, but also induces the expression of repressor protein. As long as the inducer is present in sufficient amounts, the GaaX repressor is predicted to be inactive as a repressor and thereby the GaaR transcription factor remains active. When the concentration of inducer decreases, it is reasonable to suggest that repressor proteins lacking bound inducer could inactivate the GaaR transcriptional activator, thereby restraining the expression of GA-responsive genes. Thus, high expression of the repressor could serve as a sensitive system to ensure that, when intracellular GA levels decrease, the cell can tightly turn off expression of GA-responsive genes. Moreover, this mechanism also ensures the rapid response to the presence of GA as it does not require *de novo* synthesis of GaaR. Induction simply requires the binding of inducer to the repressor and subsequent activation of GaaR via post-translational mechanisms, as the expression of GaaR is not dramatically induced by GA (Alazi et al., 2016) or in the *gaaX* mutant (this study). The expression of GA-induced genes is also controlled via CreA mediated carbon catabolite repression (de Vries *et al*. 2002; Niu *et al*. 2015). The analysis of the P*pgaX-amdS* reporter strain (Figure 2) suggests that the expression of *pgaX* is carbon catabolite repressed even in the *ΔgaaX* strain. This suggests that CreA directly represses *pgaX* expression via CreA binding sites in the *pgaX* promoter, independent of GaaX repression (Figure 6).

The proposed model for the mechanism by which GaaR and GaaX regulate gene expression resembles in some aspects the Gal3/Gal4/Gal80 module of *S. cerevisiae*, but

shows at least two important differences. Whereas the Gal4 regulatory system consists of three proteins (Gal4 as the transcriptional activator, Gal80 as the repressor and Gal3 as possible galactose sensor), we have identified two genes/proteins involved in GA regulation and no evidence for a third member. Also in the regulation of quinate metabolism, no third regulatory gene has been identified even though saturating mutant screens have been performed. These observations do not exclude the possibility that a third factor is involved in the GA or quinic acid regulation, but it is unlikely with the available evidence. Whereas the sensor (Gal3)/repressor (Gal80) function is mediated via two different proteins in the Gal regulatory system in *S. cerevisiae*, in the GA and quinic acid regulatory systems, the sensor/repressor function might well be performed by a single protein, GaaX and QutR, respectively. Another important difference is that GaaX and QutR do not show homology to Gal80 or Gal3, nor do Gal80 or Gal3 display homology to AROM. Based on these observations, we suggest that the GAL repressor module has evolved independently from that of GaaX/QutR.

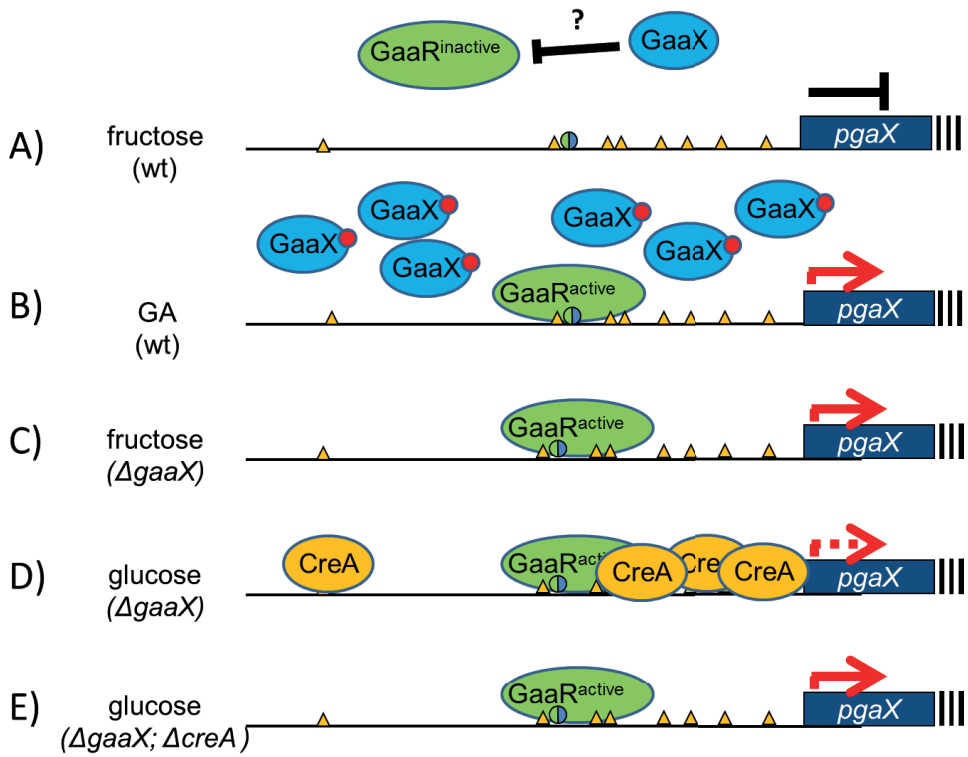

**Figure 6.** Model for the regulation of GA-induced gene expression in *A. niger.* A) GA-induced gene expression, with *pgaX* as an example, is controlled via interaction of the transcriptional activator (GaaR) and transcriptional repressor (GaaX) in combination with CreA-mediated carbon catabolite repression. A) In the presence of fructose (a non-repressible, non-inducing carbon source) *pgaX* expression is prevented because GaaX inhibits GaaR's activation . The question mark indicates that the mechanism by which GaaX controls GaaR activity is unknown. B) In the presence of GA, GA itself or a derivative of GA is predicted to bind to GaaX. The binding of the inducer to GaaX is expected to activate GaaR. GaaX is induced and remains cytosolic but the presence of the inducer keeps GaaX inactive. C) In the *ΔgaaX* strain, GaaR is no longer kept inactive by GaaX and therefore is constitutively active, resulting in constitutive expression of *pgaX*. D) In the *ΔgaaX* strain, the presence of glucose leads to CreA-mediated repression leading to reduced expression of *pgaX* and possibly other pectinolytic genes. E) Deletion of both *gaaX* and *creA* results in constitutive expression of *pgaX* even in the presence of glucose. The yellow triangles represent putative CreA binding sites. The green/blue circle represents a putative GaaR binding site. The red circle represents the postulated inducing sugar.

In addition to GaaX (NRRL3_08194) and QutR (NRRL3_11039), we identified two additional paralogues in the *A. niger* genome (NRRL3_08276 and NRRL3_07605). All four paralogues showed significant homology to the *A. niger* AROM protein as well as limited homology towards each other. Both NRRL3_08276 and NRRL3_07605 are also located next to predicted $Zn(II)_2Cys_6$ domain transcription factors, NRRL3_08275 and NRRL3_07604, respectively. Whereas the function of the GaaR/GaaX and QutA/QutR modules are related to GA and quinic acid metabolism, respectively, the function of the two other pairs that are present in *A. niger* remains to be elucidated. The sequence similarity of NRRL3_08276 and NRRL3_07605 to QutR and GaaX and their genome clustering with predicted transcription factors suggest that the proposed activator/repressor modules observed for GaaR-GaaX and QutA-QutR is an evolutionarily conserved mechanism to control gene expression in filamentous ascomycete fungi. The number of similar activator/repressor modules varies among Pezizomycotina species (Figure S4 and S5). Most Pezizomycotina species contain the galacturonic acid and quinic acid related transcriptional activator/repressor modules. It is interesting to note that some fungi, e.g. *Talaromyces stipitatus* and *B. cinerea,* seem to have lost the quinic acid specific repressor, which suggests they might have lost the capacity to utilize quinic acid. The GaaR/GaaX and QutA/QutR activator/repressor modules and their variants are specific for Pezizomycotina and missing in ascomycete yeasts, zycomycetes and basidiomycetes.

**Acknowledgements**

We thank Cees van den Hondel for helpful discussions and Tim Knetsch and Jos Reijngoud for assistance with bioreactor cultivations. Jing Niu is supported through a grant from the China Scholarship Council. Ebru Alazi is supported by a grant from BE-Basic within flagship 10 (AMBIC). This work was in part supported by Genome Canada and Génome Québec.

## References


Alazi, E., J. Niu, J. E. Kowalczyk, M. Peng, M. V. Aguilar Pontes *et al.*, 2016 The transcriptional activator GaaR of *Aspergillus niger* is required for release and utilization of D-galacturonic acid from pectin. FEBS Lett 590: 1804-1815.

Arentshorst, M., A. F. Ram and V. Meyer, 2012 Using non-homologous end-joining-deficient strains for functional gene analyses in filamentous fungi. Methods Mol Biol 835**:** 133-150.

Arentshorst M., Niu J. and A. F. Ram, 2015 Efficient Generation of *Aspergillus niger* knock out Strains by combining NHEJ mutants and a split marker approach, pp. 263-272. In Genetic Transformation Systems in Fungi, Vol. 1, edited by van den Berg MA and Maruthachalam K (eds.). Springer International Publishing.

Battaglia, E., M. Zhou and R. P. de Vries, 2014 The transcriptional activators AraR and XlnR from *Aspergillus niger* regulate expression of pentose catabolic and pentose phosphate pathway genes. Res Microbiol 165**:** 531-540.

Bennett, J.W., Lasure, L., 1991. Growth media. In: Bennett J.W., Lasure L.L. (Eds.). More gene manipulations in fungi. p441-458. Academic Press, San Diego.

Bos C.J., A.J. Debets, K. Swart, A. Huybers, G. Kobus, S.M. Slakhorst, 1988 Genetic analysis and the construction of master strains for assignment of genes to six linkage groups in *Aspergillus niger*. Curr Genet 14: 437-443.

Damveld, R. A., A. Franken, M. Arentshorst, P. J. Punt, F. M. Klis *et al.*, 2008 A novel screening method for cell wall mutants in *Aspergillus niger* identifies UDP-galactopyranose mutase as an important protein in fungal cell wall biosynthesis. Genetics 178**:** 873-881.

de Vries, R. P., J. Visser and L. H. de Graaff, 1999 CreA modulates the XlnR-induced expression on xylose of *Aspergillus niger* genes involved in xylan degradation. Res Microbiol 150**:** 281-285.

de Vries, R. P., J. Jansen, G. Aguilar, L. Parenicova, V. Joosten *et al*., 2002 Expression profiling of pectinolytic genes from *Aspergillus niger*. FEBS Lett 530: 41-47.

Dowzer, C. E., and J. M. Kelly, 1991 Analysis of the *creA* gene, a regulator of carbon catabolite repression in *Aspergillus nidulans*. Mol Cell Biol 11**:** 5701-5709.

Duncan, K., R. M. Edwards and J. R. Coggins, 1987 The pentafunctional arom enzyme of *Saccharomyces cerevisiae* is a mosaic of monofunctional domains. Biochem J 246: 375-386.

Garrison, E. and G. Marth, 2012 Haplotype-based variant detection from short-read sequencing. ArXiv:1207.3907 [q-bio.GN].

Geever, R. F., L. Huiet, J. A. Baum, B. M. Tyler, V. B. Patel *et al.*, 1989 DNA sequence, organization and regulation of the qa gene cluster of *Neurospora crassa*. J Mol Biol 207**:** 15-34.

Giles, N. H., M. E. Case, J. Baum, R. Geever, L. Huiet *et al*., 1985 Gene organization and regulation in the qa (quinic acid) gene cluster of *Neurospora crassa*. Microbiol Rev 49: 338-358.

Giles N.H., R.F.Geever, D.K. Asch, J Avalos., M.E.Case 1991 The Wilhelmine E. Key 1989 invitational lecture. Organization and regulation of the qa (quinic acid) genes in *Neurospora crassa* and other fungi. J Hered 82: 1-7.

Grant, S., C.F. Roberts, H., Lamb, M. Stout, A.R. Hawkins 1988 Genetic regulation of the quinic acid utilization (QUT) gene cluster in *Aspergillus nidulans*. J Gen Microbiol 134:3 47-58.

Gruben, B. S., M. Zhou, A. Wiebenga, J. Ballering, K. M. Overkamp *et al.*, 2014 *Aspergillus niger* RhaR, a regulator involved in L-rhamnose release and catabolism. Appl Microbiol Biotechnol 98**:** 5531-5540.

Hawkins, A. R., and M. Smith, 1991 Domain structure and interaction within the pentafunctional arom polypeptide. Eur J Biochem 196: 717-724.

Jiang, H., R. Lei, S. Ding and S. Zhu, 2014 Skewer: a fast and accurate adapter trimmer for next-generation sequencing paired-end reads. BMC Bioinformatics 15:182.

Jørgensen, T. R., B. M. Nitsche, G. E. Lamers, M. Arentshorst, C. A. van den Hondel *et al*., 2010 Transcriptomic insights into the physiology of *Aspergillus niger* approaching a specific growth rate of zero. Appl Environ Microbiol 76: 5344-5355.

Kashyap, D. R., P. K. Vohra, S. Chopra and R. Tewari, 2001 Applications of pectinases in the commercial sector: a review. Bioresour Technol 77**:** 215-227.

Khan, M., E. Nakkeeran and S. Umesh-Kumar, 2013 Potential application of pectinase in developing functional foods. Annu Rev Food Sci Technol 4**:** 21-34.

Kopylova E., L. Noé and H. Touzet, 2012 SortMeRNA: Fast and accurate filtering of ribosomal RNAs in metatranscriptomic data. Bioinformatics 28: 3211-3217.

Lamb, H. K., A. R. Hawkins, M. Smith, I. J. Harvey, J. Brown *et al.*, 1990 Spatial and biological characterisation of the complete quinic acid utilisation gene cluster in *Aspergillus nidulans*. Mol Gen Genet 223**:** 17-23.

Lamb, H. K., J. D. Moore, J. H. Lakey, L. J. Levett, K. A. Wheeler *et al.*, 1996 Comparative analysis of the QUTR transcription repressor protein and the three C-terminal domains of the pentafunctional AROM enzyme. Biochem J 313 ( Pt 3)**:** 941-950.

Langmead, B. and S. Salzberg, 2012 Fast gapped-read alignment with Bowtie2. Nature Methods 9:357-359.

Levesley, I., G. H. Newton, H. K. Lamb, E. van Schothorst, R. W. Dalgleish *et al*., 1996 Domain structure and function within the QUTA protein of *Aspergillus nidulans*: implications for the control of transcription. Microbiology 142 ( Pt 1): 87-98.

Love M. I., W. Huber and S. Anders, 2014 Moderated estimation of fold change and dispersion for RNA-seq data with DESeq2. Genome Biology 15:550.

Martens-Uzunova, E. S., and P. J. Schaap, 2008 An evolutionary conserved D-galacturonic acid metabolic pathway operates across filamentous fungi capable of pectin degradation. Fungal Genet Biol 45**:** 1449-1457.

Martens-Uzunova, E. S., and P. J. Schaap, 2009 Assessment of the pectin degrading enzyme network of *Aspergillus niger* by functional genomics. Fungal Genet Biol 46 Suppl 1**:** S170-S179.

Meyer V., M. Arentshorst , A. El-Ghezal A, A. C. Drews, R. Kooistra *et al*. 2007 Highly efficient gene targeting in the *Aspergillus niger kusA* mutant. J. Biotechnol 10:770-775.

Mohnen, D., 2008 Pectin structure and biosynthesis. Curr Opin Plant Biol 11**:** 266-277.

Nitsche, B. M., T. R. Jørgensen, M. Akeroyd, V. Meyer and A. F. Ram, 2012 The carbon starvation response of *Aspergillus niger* during submerged cultivation: insights from the transcriptome and secretome. BMC Genomics 13**:** 380.

Niu, J., T. G. Homan, M. Arentshorst, R. P. de Vries, J. Visser *et al.*, 2015 The interaction of induction and repression mechanisms in the regulation of galacturonic acid-induced genes in *Aspergillus niger*. Fungal Genet Biol 82**:** 32-42.

Niu, J., M. Arentshorst, F. Seelinger, A. F. Ram and J. P. Ouedraogo, 2016 A set of isogenic auxotrophic strains for constructing multiple gene deletion mutants and parasexual crossings in *Aspergillus niger*. Arch Microbiol 198**:** 861-868.

Patel V.B., M. Schweizer, C.C. Dykstra, S.R. Kushner, N.H. Giles. 1981. Genetic organization and transcriptional regulation in the qa gene cluster of *Neurospora crassa*. Proc Natl Acad Sci U S A 78: 5783-5787.

Patro, R., G. Duggal, M. I. Love, R. A. Irizarry, and C. Kingsford, 2016 Salmon provides accurate, fast, and bias-aware transcript expression estimates using dual-phase inference. bioRxiv 021592; doi:http://dx.doi.org/10.1101/021592

Petersen, K. L., J. Lehmbeck and T. Christensen, 1999 A new transcriptional activator for amylase genes in *Aspergillus*. Mol Gen Genet 262**:** 668-676.

Raulo, R., M. Kokolski and D. B. Archer, 2016 The roles of the zinc finger transcription factors XlnR, ClrA and ClrB in the breakdown of lignocellulose by *Aspergillus niger*. AMB Express 6**:** 5.

Richards, T. A., J. B. Dacks, J. M. Jenkinson, C. R. Thornton and N. J. Talbot, 2006 Evolution of filamentous plant pathogens: gene exchange across eukaryotic kingdoms. Curr Biol 16: 1857-1864.

Ruijter, G. J. G., and J. Visser, 1997 Carbon repression in *Aspergilli*. FEMS Microbiol Lett 151:103-114.

Sambrook J and Russell DW, 2001 *Molecular cloning: a laboratory manual*. Cold Spring Harbor Press, 3rd Edition.

Sloothaak, J., M. Schilders, P. J. Schaap and L. H. de Graaff, 2014 Overexpression of the *Aspergillus niger* GatA transporter leads to preferential use of D-galacturonic acid over D-xylose. AMB Express 4**:** 66.

Song, L., and L. Florea, 2015 Rcorrector: efficient and accurate error correction for Illumina RNA-seq reads. GigaScience 4:14.

Teather, R. M., and P. J. Wood, 1982 Use of Congo red-polysaccharide interactions in enumeration and characterization of cellulolytic bacteria from the bovine rumen. Appl Environ Microbiol 43**:** 777-780.

van Hartingsveldt, W., I. E. Mattern, C. M. van Zeijl, P. H. Pouwels, and C. A. van den Hondel, 1987 Development of a homologous transformation system for *Aspergillus niger* based on the *pyrG* gene. Mol Gen Genet 206: 71-75.

van Peij, N. N., J. Visser and L. H. de Graaff, 1998 Isolation and analysis of *xlnR*, encoding a transcriptional activator co-ordinating xylanolytic expression in *Aspergillus niger*. Mol Microbiol 27**:** 131-142.

vanKuyk P.A., J.A. Benen, H.A. Wösten, J. Visser, R.P. de Vries, 2012 A broader role for AmyR in *Aspergillus niger*: regulation of the utilisation of D-glucose or D-galactose containing oligo- and polysaccharides. Appl Microbiol Biotechnol 93: 285-293.

Yuan, X. L., R. M. van der Kaaij, C. A. van den Hondel, P. J. Punt, M. J. van der Maarel *et al.*, 2008a *Aspergillus niger* genome-wide analysis reveals a large number of novel alpha-glucan acting enzymes with unexpected expression profiles. Mol Genet Genomics 279**:** 545-561.

Yuan, X. L., J. A. Roubos, C. A. van den Hondel and A. F. Ram, 2008b Identification of InuR, a new Zn(II)2Cys6 transcriptional activator involved in the regulation of inulinolytic genes in *Aspergillus niger*. Mol Genet Genomics 279**:** 11-26.

Zhang, L., R. J. Lubbers, A. Simon, J. H. Stassen, P. R. Vargas Ribera *et al.*, 2016 A novel Zn2Cys6 transcription factor BcGaaR regulates D-galacturonic acid utilization in *Botrytis cinerea*. Mol Microbiol 100**:** 247-262.

## Supporting information

The supplementary material of this chapter are available via

http://www.genetics.org/content/205/1/169.figures-only and comprises the following:

**Table S1.** Primers used in this study.

**Table S2.** SNPs and indels detected in genomes of mutants.

**Table S3.** (A) TPM values of NRRL3 predicted ORFs (NRRL3_00001-NRRL3_11846) and (B) differentially expressed genes between WT and Δ*gaaX*.

**Table S4.** Expression data of genes proposed to be part of the GaaR-GaaX panregulon.

**Table S5.** Synteny of the GaaR-GaaX orthologs among 17 *Pezizomycotina*.

**Figure S1.** Verification of gaaX deletion strains in the JC1.5 and JN29.2 backgrounds.

**Figure S2.** Verification of the gaaX deletion strain in the MA323.1 background and targeted integration of the GFP fusion constructs at the pyrG locus.

**Figure S3.** Strategy and primer design for construction of GaaX-GFP fusion proteins.

**Figure S4.** Schematic overview of the presence or absence of GaaX-like repressor proteins in the genomes of 17 *Pezizomycotina* species, as well as S. cerevisiae, and representative Zygomycotina (*Mucor circinelloides*) and Basidiomycotina (*Ustilago maydis*).

**Figure S5.** Phylogenetic analysis of fungal AROM and AROM-related repressor proteins in 17 Pezizomycotina species, as well as S. cerevisiae, and representative Zygomycotina (*Mucor circinelloides*) and Basidiomycotina (*Ustilago maydis*).