X-linked susceptibility to mycobacteria is caused by mutations in NEMO impairing CD40-dependent IL-12 production.


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Germline mutations in five autosomal genes involved in interleukin (IL)-12–dependent, interferon (IFN)–γ–mediated immunity cause Mendelian susceptibility to mycobacterial diseases (MSMD). The molecular basis of X-linked recessive (XR)–MSMD remains unknown. We report here mutations in the leucine zipper (LZ) domain of the NF-κB essential modulator (NEMO) gene in three unrelated kindreds with XR–MSMD. The mutant proteins were produced in normal amounts in blood and fibroblastic cells. However, the patients’ monocytes presented an intrinsic defect in T cell–dependent IL-12 production, resulting in defective IFN-γ secretion by T cells. IL-12 production was also impaired as the result of a specific defect in NEMO- and NF-κB/c-Rel–mediated CD40 signaling after the stimulation of monocytes and dendritic cells by CD40L–expressing T cells and fibroblasts, respectively. However, the CD40L-dependent up-regulation of costimulatory molecules of dendritic cells and the proliferation and immunoglobulin class switch of B cells were normal. Moreover, the patients’ blood and fibroblastic cells responded to other NF-κB activators, such as tumor necrosis factor–α, IL-1β, and lipopolysaccharide. These two mutations in the NEMO LZ domain provide the first genetic etiology of XR–MSMD. They also demonstrate the importance of the T cell– and CD40L–triggered, CD40L–, and NEMO/NF-κB/c-Rel–mediated induction of IL-12 by monocyte–derived cells for protective immunity to mycobacteria in humans.
Mendelian susceptibility to mycobacterial diseases (MSMD) (MIM 209950) is a congenital syndrome resulting in predisposition to clinical disease caused by weakly virulent mycobacterial species, such as BCG vaccines and nontuberculous, environmental mycobacteria (EM) (1–4). Patients are also susceptible to the more virulent species *Mycobacterium tuberculosis* (5). Diseases caused by nontyphoidal *Salmonella* serotypes have been observed in just under half the cases (4). Other infectious diseases have only rarely been reported, mostly in single patients. A few patients have suffered from viral infections, including cytomegalovirus and human herpes virus-8 infections, and others have had fungal infections with species such as *Histoplasma capsulatum* and *Paracoccidioides brasiliensis*. MSMD was initially thought to be autosomal recessive in most, if not all kindreds, as a result of the high frequency of both multiple-case sibships and consanguineous kindreds (2, 3).

Figure 1. Clinical phenotypes and NEMO genotypes of patients. (A) Pedigrees of three kindreds with NEMO mutations (A, B, and C). Each generation is designated by a Roman numeral, and each individual is designated by an Arabic numeral. Male patients (in black squares) present mycobacterial infections and hemizygous NEMO mutations; asymptomatic female carriers are represented by black dots. The probands are indicated by arrows. (B) The teeth of patient P1 (Kindred A, II.4, sparse teeth), patient P2 (Kindred B, II.1, conical decidual incisors), and patient P3 (Kindred C, II.1, normal teeth). (C) Sequence electrophoregram of NEMO complementary DNA in the region corresponding to the mutation. Kindred A, P1 presents the mutation A→C leading to the replacement at residue 315 of Glu (E) by Ala (A) (E315A). Kindred B, P2 and kindred C, P3 present the mutation G→A resulting in the replacement of an Arg (R) at position 319 by Gln (Q) (R319Q). (D) Schematic representation of the NEMO coding region, from exons 2 to 10, and corresponding domains, shown in dark gray: the coil-coiled domain 1 (CC1, Leu 93 to Gln 194) and coil-coiled domain 2 (CC2, Met 258 to Lys 292), and the leucine zipper (LZ, Tyr 308 to Lys 344) and zinc finger (ZF, Cys 397 to Cys 417) domains. All published hypomorphic NEMO mutations (associated with the various forms of EDA-ID) are represented (reference 43). Missense and nonsense mutations (amino-acid code) are shown on top, whereas splice and frameshift mutations (nucleotide code) are shown on the bottom. NEMO mutations in red represent mutations associated with mycobacterial infections.
After the identification in 1996 of germline mutations in IFNGR1, encoding the IFN-γ receptor ligand-binding chain (IFN-γR1) (6, 7), recessive mutations were reported in three other autosomal genes: IFNGR2, encoding the accessory chain of the IFN-γ receptor (8); IL12B, encoding the p40 subunit shared by IL-12 and IL-23 (9); and IL12RB1, encoding the β1 chain shared by the receptors for IL-12 and IL-23 (10, 11). Defects in IFNGR1 and IFNGR2 are associated with impaired cellular responses to IFN-γ, whereas defects in IL12B and IL12RB1 are associated with impaired IFN-γ production. Allelic heterogeneity at these four loci accounts for the existence of nine autosomal recessive disorders. After the identification of complete defects in which no protein is expressed (6–8, 10, 11), partial forms of IFN-γR1 (12) and IFN-γR2 (13) deficiency and complete forms of IFN-γR1 (14), IFN-γR2 (15), and IL-12Rβ1 (16) deficiency with surface receptor expression were identified.

The identification of MSMD-causing recessive alleles in these four autosomal genes led to the discovery of an autosomal dominant form of MSMD, caused by dominant-negative mutations in these four autosomal genes led to the discovery of an autosomal dominant form of MSMD, caused by dominant-negative mutations in these four autosomal genes. After the identification of complete defects in which no protein is expressed (6–8, 10, 11), partial forms of IFN-γR1 (12) and IFN-γR2 (13) deficiency and complete forms of IFN-γR1 (14), IFN-γR2 (15), and IL-12Rβ1 (16) deficiency with surface receptor expression were identified.

The identification of MSMD-causing recessive alleles in these four autosomal genes led to the discovery of an autosomal dominant form of MSMD, caused by dominant-negative alleles of IFNGR1 (17). The mutant alleles exert a dominant-negative effect as the result of overexpression, at the cell surface, of truncated chains that cannot transduce signals. Dominant mutations in STAT1, another autosomal gene encoding the signal transducer and transactivator of transcription 1 (Stat-1), were subsequently identified (18). There are two forms of partial Stat-1 deficiency, depending on whether the mutation impairs the phosphorylation of Tyr 701 (18) or DNA-binding activity (unpublished data). The two forms of partial Stat-1 deficiency are associated with MSMD because these mutations affect the activation of IFN-γ–induced Stat-1 homodimers, but not of IFN-α/β–induced Stat-1–Stat-2–IRF-9 trimers, in heterozygous cells. A dominant-negative mutant allele of IFNGR2 has also been identified, resulting in the aberrant subcellular distribution of IFN-γR2, as the result of a mutation affecting the transmembrane domain (19).

In one family first reported in 1991, and more extensively in 1994, it was suggested that MSMD might display X-linked recessive inheritance (1, 20–22). Four maternally related male patients were found to suffer from severe EM disease. These patients’ monocytes were found to be intrinsically unable to produce normal levels of IL-12 upon stimulation by PHA and T cells (21, 22). Despite the identification of this immunological phenotype, the molecular basis of XR-MSMD has remained elusive. We report here the identification of MSMD-causing NEMO mutations in this kindred and in two other, unrelated kindreds. These mutations do not affect NF-κB activation in response to most classical activators tested, accounting for the narrow spectrum of infections in patients. The two NF-κB essential modulator (NEMO) mutations identified selectively impair the CD40–triggered and NF-κB/c-Rel–mediated induction of IL-12 production by monocytes and monocyte-derived dendritic cells, accounting for the susceptibility to mycobacteria.

RESULTS

New germline mutations in NEMO in three kindreds

We studied an American multiplex kindred, a French kindred, and a German kindred, each containing one male patient with sporadic mycobacterial disease (Fig. 1 A). None of the six patients in these kindreds displayed the classical developmental and infectious features of anhidrotic ectodermal dysplasia with immunodeficiency (EDA-ID) (23–25). Coni cal incisors in one proband (P2, kindred B, Fig. 1 B) and in a second patient (patient III.8, kindred A) nevertheless led us to study the X-linked EDA-ID–causing NEMO gene (26) (unpublished data). New mutations were found in the coding region of NEMO in the three probands (Fig. 1 C). P1 from kindred A presented a nucleotide substitution at position 944 (A→C) (Fig. 1 C, left), leading to the replacement of a Glu by an Ala at residue 315 (E315A) (Fig. 1 D). P2 and P3 from kindreds B and C, respectively, presented a nucleotide substitution at position 956 (G→A) (Fig. 1 C, middle).
and right) resulting in the replacement of an Arg by a Gln at position 319 (R319Q) (Fig. 1 D). The two mutations were detected in cDNAs, indicating that they corresponded to NEMO and not the nearby NEMO pseudogene (27). These mutations were not found in 800 unrelated healthy controls (1,200 X chromosomes) from 51 ethnic groups (HGDP-CEPH database). The familial segregation of NEMO in kindred A was consistent with linkage between the observed syndrome and this gene (unpublished data). The heterozygous grandmother (I.1), and three other women in kindred A (II.3, III.2, III.10), like the mother of P2 in kindred B (I.1) and the mother of P3 in kindred C (I.1), were obligate carriers, but displayed no signs of incontinentia pigmenti or EDA (unpublished data). These data suggest that these three unrelated families suffer from XR-MSMD as the result of missense mutations in NEMO.

Modeling of the molecular environment of the E315 and R319 residues
Amino acids E315 and R319 are both located within the leucine zipper (LZ) domain of NEMO (Fig. 1 D). Several studies have shown that NEMO self-assembles into trimers (28, 29). The minimal oligomerization domain of the protein consists of the CC2 and LZ coiled-coil motifs, and we recently suggested that these motifs interact to form a pseudo-six-helix bundle, in which the LZ α-helices dock in the crevices defined by two of the helices of the central trimeric CC2 coiled-coil (Fig. 1 A) (30). A similar scaffold was described for the HIV-1 gp41 protein (31). We therefore modeled the NEMO trimeric CC2-LZ domain based on the X-ray structure of the gp41 ectodomain (Fig. 2 B). In our model, the E315 and R319 residues, located in the middle of the LZ domain, face the interior of the coiled coil assembly. The environment of the two residues is shown in greater detail in Fig. 2 C. E315 and R319 probably form an intramolecular salt bridge stabilizing the LZ α-helix. Other amino acids located in the vicinity of these residues in the model are also shown, including K277, from the same NEMO subunit, and Q278 from the CC2 motif of an adjacent subunit, which probably interact via a hydrogen bond. The E315A and R319Q mutations found in XR-MSMD patients would break the predicted salt bridge and might induce local plasticity in the LZ helices of the NEMO trimer. Within the highly structured CC2-LZ trimer, this might also result in local modifications to interactions with the central CC2 domains. Our structural model therefore also provides a rationale for the remarkable similarity and narrowness of the clinical phenotypes of the patients described, with NEMO mutations disrupting a common salt bridge in the LZ domain.

Normal expression of NEMO protein in blood cells and fibroblasts
We assessed NEMO expression by Western blotting extracts of EBV-transformed B cells and SV40-transformed fibroblasts with p18 and 3328 antibodies, which recognize the N-terminal region of NEMO. The mutant proteins, E315A (P1) (not depicted) and R319Q (P2), were expressed in similar amounts to wild-type NEMO in EBV-transformed B cells (Fig. 3 A). The R319Q protein was also expressed in normal amounts in SV40-transformed fibroblasts from P2 (Fig. 3 B). Cell lines from P3 (R319Q) and fibroblasts from P1 (E315A) were not available. The NEMO X420W mutant, which causes the most severe form of EDA-ID (24), with osteopetrosis and lymphoedema (OL-EDA-ID), was detected only in trace amounts, at a higher molecular weight, on Western blots of EBV-transformed B cells and SV40-transformed fibroblasts (Fig. 3, A and B). As a negative control (C−), no NEMO was detected in SV40-transformed fibroblasts bearing a large amorphic deletion in NEMO (27). We assessed intracellular NEMO expression by flow cytometry, using antibody 278–396, which recognizes the C-terminal region of NEMO (32). NEMO was normally detected by Western blotting with two specific antibodies, 3328 and p18, which recognize different parts of the N-terminus of NEMO (A) EBV-transformed B cell lines and (B) SV-40-transformed fibroblastic cell lines from a healthy donor (C+), an OL-EDA-ID patient carrying the X420W NEMO mutation, P2 and an SV-40-transformed fibroblastic cell line from a NEMO-deficient fetus as a negative control (C−). Intracellular staining of NEMO with a specific antibody recognizing amino acids 278–396 and detected by flow cytometry in (C) EBV-transformed B cell lines and (D) SV40-transformed fibroblastic cell lines from healthy control (C+), an OL-EDA-ID patient, P1 and P2. (E) Purified monocytes (left) and T cells (right) from a healthy control (C+) and P2.

Figure 3. Expression of NEMO in the patients. NEMO protein, detected by Western blotting with two specific antibodies, 3328 and p18, which recognize different parts of the N-terminus of NEMO (A) EBV-transformed B cell lines and (B) SV-40-transformed fibroblastic cell lines from a healthy donor (C+), an OL-EDA-ID patient carrying the X420W NEMO mutation, P2 and an SV-40-transformed fibroblastic cell line from a NEMO-deficient fetus as a negative control (C−). Intracellular staining of NEMO with a specific antibody recognizing amino acids 278–396 and detected by flow cytometry in (C) EBV-transformed B cell lines and (D) SV40-transformed fibroblastic cell lines from healthy control (C+), an OL-EDA-ID patient, P1 and P2. (E) Purified monocytes (left) and T cells (right) from a healthy control (C+) and P2.
expressed in EBV-transformed B cells (P1, P2) and SV40-transformed fibroblasts (P2) from the patients, taking seven and five control cell lines, respectively, as a reference, but not in fibroblasts bearing a large deletion in NEMO (Fig. 3, C and D). NEMO was barely detectable in EBV-transformed B cells and SV40-transformed fibroblasts from the patient with OL-EDA-ID. The intracellular expression of NEMO was also normal in freshly isolated monocytes and T cells from P2 (Fig. 3 E). Thus, levels of NEMO expression were strictly normal in patients bearing the E315A and R319Q mutations as shown by Western blotting and flow cytometry in hematopoietic and nonhematopoietic cell lines and in freshly prepared blood cells.

**Response of blood cells and fibroblasts to LPS, R-848, TNF-α, and IL-1β**

NEMO mutations in patients with EDA-ID are usually associated with impaired cellular responses to multiple stimuli, including LPS (via TLR-4), TNF-α, and IL-1β (24, 25). We thus stimulated whole blood cells from the patients and from 

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**Figure 4. NF-κB activation in leukocytes and fibroblasts.**

Healthy controls (C+, ●), OL-EDA-ID patient (△), SV-40-transformed fibroblastic cell line from NEMO-deficient fetus (C−, ▲), P2 (□), and P3 (○). (A) IL-10 production by whole blood cells from 10 healthy controls and P2 and P3, upon stimulation with LPS (1 μg/ml), TNF-α (20 ng/ml), IL-1β (10 ng/ml), and PMA/ionomycin (10−7/10−5 M) for 48 h. (B) TNF-α production by EBV-transformed B cells after 24 h of TLR7/8 activation by R-848 agonist. (C) IL-6 and IL-8 production by SV-40-transformed fibroblastic cell lines from nine healthy controls and from P2 and C− (three experiments each for P2 and C−), after 24 h of TNF-α and IL-1β stimulation. All cytokines were determined by sandwich ELISA. (D) DNA-binding activity in nuclear extracts from fibroblastic cell lines after TNF-α (20 ng/ml) and IL-1β (10 ng/ml) activation for various lengths of time, as detected with a radio-labeled DNA probe by EMSA (top), and κBα degradation and GAPDH expression, detected by Western blotting, in the corresponding cytoplasmic extracts (bottom). The results shown are representative of at least two independent experiments.
10 healthy controls with LPS, TNF-α, IL-1β, and PMA/ionomycin, and measured the induction of IL-6 (unpublished data), IL-10, and TNF-α. The three patients displayed normal levels of IL-10 (Fig. 4 A), IL-6, and TNF-α (not depicted) production upon stimulation with LPS, IL-1β, and PMA plus ionomycin. IL-10 was also induced by TNF-α in P2 and P3, although to lower levels (Fig. 4 A). EBV-transformed B cells from P2 responded normally to R-848, an agonist of TLR-7/8, in terms of TNF-α production (Fig. 4 B). We then assessed the impact of NEMO mutations in fibroblasts from P2, a NEMO-deficient fibroblastic cell line bearing a large NEMO deletion, and fibroblasts from nine healthy controls. Fibroblasts from P2 produced normal amounts of IL-6 and IL-8 in response to both IL-1β and TNF-α (Fig. 4 C). In contrast, fibroblasts from all EDA-ID patients tested, bearing other NEMO mutations, showed various levels of impairment of IL-6 and IL-8 production (unpublished data). We assessed IkBα cytoplasmic degradation by Western blotting and the κB-binding activity of nuclear extracts by electrophoretic mobility shift assays. In contrast with what was observed in fibroblasts from the OL-EDA-ID patient bearing NEMO mutation X420W (24), both IkBα degradation and κB binding activity were completely normal in fibroblasts from P2 (Fig. 4 D). Thus, the NEMO–NF-κB signaling pathway in response to TLR4, 7, and 8 agonists, IL-1β, and TNF-α, was normal in SV40-fibroblastic and EBV-transformed B cell lines from our patients.

Impaired production of IL-12 and IFN-γ by peripheral blood mononuclear cells upon PHA activation

We previously reported that PBMCs from P1 produced small amounts of IFN-γ upon stimulation with PHA (21, 22). PBMCs from P2 and P3 produced normal amounts of IFN-γ in response to PMA/ionomycin but not in response to PHA or CD3 stimulation, with normal levels determined as those for 10 healthy controls (Fig. 5 A). In these conditions, the impaired production of IL-12 was previously shown to account for poor IFN-γ production by PBMCs in P1 (21), consistent with the poor production of IFN-γ observed in blood cells from IL-12p40- and IL-12Rβ1-deficient patients (9, 11, 33). Accordingly, PBMCs from P2 and P3 produced only small amounts of IL-12 after PHA stimulation, which were not complemented by adding IFN-γ (Fig. 5, B and C). In contrast, the addition of IL-12 partially complemented IFN-γ production (Fig. 5 A). As a control, the PHA-driven production of TNF-α and IL-10 by P2 and P3 was similar to that in 10 healthy controls (unpublished data). The production of IL-12 and IL-12β-γ was also normal upon stimulation with LPS and PMA/ionomycin, respectively (Fig. 5, A–C). Moreover, the production of IL-12 and IFN-γ by whole blood cells stimulated with live BCG (alone or with IFN-γ or IL-12) was normal in P2 and P3 (Fig. 5 D). Thus, the three probands displayed selective impairment of IL-12 and IFN-γ production by PBMCs upon stimulation with PHA or CD3. In contrast, the production of IL-12 was normal in response to LPS and BCG and that of IFN-γ was normal in response to PMA/ionomycin and BCG. Overall, the cellular phenotype of impaired IL-12–dependent production of IFN-γ by PHA-stimulated PBMCs is suggestive of a common pathogenic mechanism of predisposition to mycobacterial disease associated with NEMO mutations E315A and R319Q.

Impaired IL-12 secretion by PHA-stimulated monocytes and T cells

We previously described a specific defect in IL-12 production by P1 monocytes upon PHA stimulation in the presence of control T cells (21, 22). We tested cells from P2 and P3 in a similar coculture assay and measured the production of IL-12p70, IL-12p40, and IFN-γ by ELISA (Fig. 6 A). These cytokines were produced in very small amounts or not at all in monocytes stimulated with PHA in the absence of T cells or in T cells in the absence of monocytes. P2 and P3 monocytes cocultured with autologous or healthy T cells produced much less IL-12p70, IL-12p40, and IFN-γ than control monocytes cocultured with autologous, P2, or P3 T cells. The T lymphocytes of these patients produced IFN-γ after costimulation with control monocytes and PHA. The low levels of IFN-γ secretion were the result of poor IL-12 production as indicated by (a) the complementation provided by recombinant IL-12 (but not IL-23) (unpublished data) and (b) the poor production of IFN-γ documented with IL-12p40–deficient monocytes or IL-12Rβ1-deficient

Figure 5. IL-12 and IFN-γ production by leukocytes. Cytokine production by PBMCs from 10 healthy donors, (C+, ●), P2 (□), and P3 (○) in response to PHA, alone or in combination with recombinant IL-12 (20 ng/ml), IL-23 (20 ng/ml), or IFNγ (5,000 U/ml), anti-CD3 (10 ng/ml), LPS (1 μg/ml) and PMA/ionomycin (10−7/10−5 M). (A) IFN-γ, (B) IL-12p70, (C) IL-12p40 production. (D) Cytokine production by whole blood cells from 50 healthy donors, P2, and P3 upon stimulation with live BCG alone or in combination with IL-12 (20 ng/ml) or IFN-γ (5,000 U/ml): IFN-γ (top), IL-12p70 (middle), and IL-12p40 (bottom) secretion. For each patient, the results shown are representative of two independent experiments.
cytokines secreted by T cells and monocytes (TNF-α, IL-7, IL-10, and MCP-1) were poorly induced, whereas others were induced in normal (IL-4, IL-10) or large (IL-5, IL-13) amounts by monocytes from P2 and P3, indicating that these cells were not globally unresponsive (Fig. 6 B and not depicted). Two other lymphokines (IL-2, IL-4) were not affected and the production of IL-17 was impaired. Finally, cytokines secreted by T cells and monocytes (TNF-α, IL-7, IL-10, and GM-CSF) were normally induced. All three probands had a specific immunological phenotype in which IL-12 production by monocytes in response to PHA and T cells was affected. Impaired IFN-γ production does not reflect an intrinsic T cell defect and is instead a consequence of impaired IL-12 production by monocytes.

**PHA-activated T cell–dependent monocyte secretion of IL-12 is driven by CD40**

The two NEMO mutations exerted their effects downstream from monocyte receptors triggering the induction of IL-12. The CD40 signaling pathway has been shown to be critical for the T cell–dependent induction of IL-12 in monocytes and dendritic cells (34–37). After PHA stimulation, PBMCs from CD40- and CD40L-deficient patients (38) produced much lower levels of IL-12 and IFN-γ than did PBMCs from 10 healthy controls (unpublished data), consistent with previous data (39). The induction of IL-12 by LPS and that of IFN-γ by PMA/ionomycin were normal (unpublished data). IL-12 induction was abolished if CD40L-deficient T cells were cocultured with healthy or CD40L-deficient monocytes, resulting in low levels but not the total absence of IFN-γ production (Fig. 7 A). Conversely, normal IL-12 induction was observed if control or CD40L-deficient monocytes were mixed with T lymphocytes from a healthy donor. Low levels of IFN-γ induction were associated with the presence of small amounts of IL-12 in this assay. IL-12 induction was abolished if CD40L-deficient monocytes were incubated with CD40L-deficient or normal T cells (Fig. 7 B). Paradoxically, IFN-γ secretion was not significantly impaired, perhaps reflecting the direct cross-linking of CD40L by PHA (40). In contrast, control monocytes produced normal amounts of IL-12 when cultured with control or CD40L-deficient T cells. Our findings for the monocyte–T cell coculture assay unambiguously indicate that the engagement of the monocyte CD40 by CD40L-expressing T cells is required for the optimal induction of IL-12 production. These data suggest that the NEMO mutations in P1, P2, and P3 were pathogenic as a result of their impact on CD40 signaling within monocytes, leading to low levels of IL-12 secretion upon T cell stimulation.

**NEMO mutation impairs CD40 signaling in dendritic cells but not B cells**

We assessed the responses to CD40 stimulation of cells from our patients. We first incubated purified monocytes from P2 with mouse fibroblastic L cells (L-cells) and L-cells stably transfected with a construct encoding human CD40L (L-cells-hCD40L). Monocytes from P2 did not produce IL-12, IL-6, and TNF-α upon stimulation with L-cells-hCD40L (unpublished data). We then tested monocyte-derived dendritic cells (MDDCs) as DCs are major producers of IL-12 in vivo. MDDCs produce large amounts of IL-12 upon CD40 stimulation in vitro (36). MDDCs from P2 and P3 were activated with LPS plus IL-1β, L-cells, or L-cells-hCD40L with or without IL-1β. MDDCs from P2 and P3 displayed severely impaired cytokine production in response to L-cells-hCD40L but not in response to LPS plus IL-1β (Fig. 7 C), whereas induction of the costimulatory molecules...
CD80 and CD86, unlike that of CD40, was maintained (Fig. 7 D). These data are consistent with our previous observations in a child with a severe NEMO mutation associated with OL-EDA-ID and severe mycobacterial infection (24). MDDCs from P2 and P3 expressed normal levels of NEMO (Fig. 7 E). B cells from P2 produced normal amounts of IgE in response to costimulation with IL-4 and CD40L, as shown by ELISA, with data from 20 healthy controls and a CD40-deficient patient used as a reference (Fig. 7 F). Moreover, B cell proliferation, as assessed by the measurement of [3H] thymidine incorporation, was also normal (unpublished data). These data suggest that the CD40 signaling pathway was not affected in B cells in vitro, consistent with the normal B cell switch and antibody response in vivo in our patients. Altogether, these findings suggest that the patients with XR-MSMD bearing the E315A and R319Q mutations in NEMO suffered from mycobacterial disease as a result of the impaired IL-12 production by monocytes and dendritic cells after stimulation with the CD40L on T cells.

Delayed nuclear accumulation of NF-κB/c-Rel in CD40-stimulated dendritic cells

We evaluated the impact of the NEMO mutations identified by confocal microscopy analysis of the subcellular distributions.
of the NF-κB subunits RelA (p65) and c-Rel in MDDCs from P2 and a healthy control, unstimulated and in response to LPS, IL-1β, TNF-α, mouse L-cells, or human L-cells expressing mouse L-cells. LPS treatment resulted in the nuclear accumulation of RelA and c-Rel in MDDCs from both the healthy control and P2 (Fig. 8 A). IL-1β induced the translocation of RelA, but not of c-Rel, in MDDCs from both the healthy donor and P2 (unpublished data). The nuclear translocation of RelA in response to TNF-α seemed to be less efficient in MDDCs from P2 (Fig. 8 A), possibly accounting for the weak induction of IL-10 in PBMCs (Fig. 4 A). The translocation of RelA in response to CD40 stimulation was normal in MDDCs from P2 up to 8 h after stimulation (Fig. 8 B). In contrast, the nuclear accumulation of c-Rel in response to CD40 was clearly impaired in MDDCs from P2, 2 (Fig. 8 B) and 5 h after stimulation (not depicted). Only after 8 h of stimulation was c-Rel accumulation detected in the nuclei of MDDCs from P2 (Fig. 8 B). These data strongly suggest that the NEMO mutation identified in P2 impairs CD40-triggered NF-κB activation by delaying the nuclear accumulation of c-Rel. They also suggest that CD40-driven, NEMO-dependent IL-12 production in human MDDCs depends on c-Rel, as previously shown in mice (41, 42). Furthermore, our data indicate that the R319Q and E315A mutations in the NEMO LZ domain are associated with XR-MSMD because they impair the T cell–dependent, CD40-triggered, NF-κB/c-Rel–mediated induction of IL-12 in monocytes and dendritic cells (Fig. 8 C).
**DISCUSSION**

We describe here the first genetic etiology of XR-MSMD, a syndrome clinically defined for the first time in 1994 in a multiplex American kindred (1, 21), after the publication of individual case reports (20). The four patients from this family carried the E315A mutation in NEMO. XR-MSMD is not limited to a single family, as a different, but related NEMO mutation, R319Q, was found in two unrelated European boys, from France and Germany. NEMO is the sixth MSMD-causing gene to be identified, after the identification of IFNγR1 in 1996 (6, 7), IFNγR2, IL12B, and IL12RB1 in 1998 (8–11), and STAT1 in 2001 (18) (Fig. 8 C). It is the sixth genetic defect associated with MSMD but the 13th genetic etiology of this syndrome, given the considerable allelic heterogeneity of previously reported defects of the IL-12–IFN-γ circuit (15) (unpublished data). One of our patients with EM disease (II.4 in kindred A) also suffered from miliary tuberculosis in childhood, and two of our probands (P2 and P3) probably suffered from tuberculosis, as the sole infectious phenotype suggested by consistent histological results and strongly positive tuberculin skin tests. Patients with BCG and EM diseases and otherwise healthy children with severe tuberculosis should therefore be investigated for NEMO mutations (5). The possibility of NEMO mutations cannot be excluded by the absence of developmental signs, as shown in four of our six patients and in two other recently reported patients (43).

We can now suggest a rationale for the pathogenesis of mycobacterial diseases in patients with XR-MSMD. The two causal NEMO mutations impair CD40 signaling in monocytes and dendritic cells, delaying the nuclear accumulation of NF-κB/c-Rel and impairing IL-12 secretion upon stimulation by CD40L-expressing T cells. In turn, impaired IL-12 production by antigen-presenting cells results in impaired IFN-γ production by T cells. X-linked NEMO is thus physiologically connected to the five autosomal IL-12/23–IFN-γ circuit genes known to cause MSMD (4). These data are consistent with the observation that CD40-deficient (44) and CD40L-depleted mice (45) are susceptible to *Mycobacterium avium* infections. CD40L-deficient mice are also susceptible to BCG and CD40-deficient mice are susceptible to *Mycobacterium tuberculosis* (46, 47). Moreover, CD40L-deficient patients frequently develop localized disease caused by BCG, and severe tuberculosis (5, 48). However, human CD40 and CD40L are not recognized as bona fide MSMD-causing genes, as CD40- and CD40L-deficient patients have never been reported to suffer from disseminated BCG or EM disease. This implies that pathways other than CD40–NEMO–IL-12, such as the TNF-α signaling pathway (Figs. 4 A and 8 A) (49), are probably involved in the pathogenesis of diseases caused by EM, at least, in patients with mutations in the NEMO LZ domain. In any event, human phagocytes and dendritic cells require T cell stimulation via CD40, NEMO, and c-Rel to produce the amounts of IL-12 necessary to control mycobacteria in natural conditions of infection (50).

Null mutations in NEMO are lethal in utero in male fetuses (27). Patients with hypomorphic mutations in NEMO suffer from XR-EDA-ID (23–26). To date, 36 patients bearing 28 different NEMO mutations have been described (23–27, 32, 43, 51–55) (unpublished data). 11 out of the known 36 XR-EDA-ID patients have developed severe mycobacterial infection, mostly caused by *M. avium*. However, these patients also suffered from infections caused by many other microorganisms. In contrast, five of our six patients presented mycobacterial disease as their sole invasive infection. The invasive *Haemophilus influenzae* b infection in one of our patients may not have occurred by chance. Similarly, two out of the six patients had conical incisors (decidual teeth in one and permanent teeth in the other), implying that the penetrance of these particular NEMO genotypes was low (but not null) for pyogenic infections and developmental defects, but high for mycobacterial disease. Our report stresses the heterogeneity of phenotypes associated with hypomorphic NEMO mutations. Two patients with XR-EDA-ID, both with the X420W genotype, presented XR-OL-EDA-ID (24, 53, 54). Most patients present the less severe clinical syndrome of XR-EDA-ID, with typical (23–25) or hidden (conical incisors) (26) developmental signs and multiple infections. Two other patients with severe infections but no developmental disorder were recently reported (43). Finally, we report here two novel NEMO mutations, responsible for MSMD in six patients. All six patients are selectively susceptible to mycobacteria and display selective impairment of the CD40-dependent induction of IL-12 in antigen-presenting cells. These findings provide the first link between a NEMO genotype and immunological and infectious phenotypes.

Why do these two mutations have selective effects? The E315A and R319Q mutations are located in the LZ domain of NEMO and do not affect protein expression. Residues E315 and R319 probably form a salt bridge that is disrupted by the two pathogenic mutations (Fig. 2 C). The two mutations may induce local plasticity in a region of the LZ domain critical for signaling via CD40, but less important for other signaling pathways. However, CD40 signaling was not totally abolished in dendritic cells from our patients, as the nuclear accumulation of RelA, and the induction of CD80 and CD86 were normal, whereas the induction of CD40 was not. These findings are consistent with the normal CD40-mediated induction of HLA-II and CD86 previously reported in dendritic cells from a patient with OL-EDA-ID (24). It is unknown whether the CD40–CD80/86 and CD40–HLA-II pathways are truly NEMO independent or whether they are unaffected by the NEMO mutations tested. The E315A and R319Q NEMO mutations impair at least one CD40 signaling pathway involving NF-κB–c-Rel in monocytes and dendritic cells, but they have no significant effect on B cells, in vivo and in vitro, unlike other previously reported NEMO mutations (24, 25, 52, 55). In conclusion, specific NEMO mutations may selectively target the CD40–NF-κB–c-Rel signaling pathway in monocytes and dendritic
cells, preventing the production of sufficient quantities of IL-12 after T cell stimulation. This results in impaired IFN-γ–mediated immunity and predisposition to mycobacterial diseases (Fig. 8 C).

MATERIALS AND METHODS
Case reports. The first family (kindred A) with XR-MSMD has been described elsewhere (1, 20–22). None of the family members were vaccinated with BCG. Severe *M. avium* infection was documented in four maternally related male family members in two successive generations (Fig. 1 A; patients II.1, II.4, III.7, and III.8). The clinical features of patient 1 (P1, II.4), the index case, have been described elsewhere (1, 20). In brief, at the age of 13 yr, P1 presented granulomatous cutaneous lesions, thought to be sarcoid. Extensive erosive lesions on the face and arm were subsequently found to be the result of *M. avium* complex infection. Treatment with rifampicin, ethambutol, clofazimine, isoniazid and streptomycin was initiated. After 2.5 yr of therapy, smears and cultures of skin from the patient’s face and arm remained positive for *M. avium* complex X cluster. Intensive antibiotic and IFN-γ therapy has led to periods in which the skin has healed and cultures have tested negative, but this patient has frequently displayed intercurrent mycobacteremia over the last 10 yr. Three other family members also developed disseminated *M. avium* complex infection. Patient II.1 was cured of miliary tuberculosis at the age of 6 yr. Disseminated *M. avium* infection occurred in this patient at the age of 40 yr, and was never completely eradicated. The patient died of *Enterobacter* bacteremia complicating parenteral nutrition at the age of 48 yr. Patient III.7 was successfully treated for disseminated *M. avium* infection at the age of 5 yr, but died in an automobile accident at the age of 10. His brother, patient III.8, had recurrent *Haemophilus influenzae* bacteraemia at the age of 6 yr. At the age of 14 yr, he presented disseminated *M. avium* complex infection involving abdominal lymph nodes and blood. Overt signs of ectodermal dysplasia (conical teeth, hypodontia, hypotrichosis, abnormal hair whorl) were evaluated. This patient has remained mycobacteremic despite treatment with multiple antibiotics and IFN-γ. Sparse teeth were the only developmental abnormality on physical examination, and long went unrecognized in patient 1 (Fig. 1 B, left). Cells of the various blood lineages and lymphoid subsets (CD3, CD4, CD8, CD20, CD16, CD56) were present in normal numbers. T cells proliferated normally in vitro in response to mitogenic (PHA) and antigenic (recall antigens) stimulation. The results of NBT and dihydroorhodamine tests were normal. Serum levels of the immunoglobulin IgG, IgM, and IgD isoforms were high (maximum titers: IgG 70.85 g/liter, IgM 1.54 g/liter, and IgD 304 IU/ml). Serum IgA and IgE levels were normal. There was a normal Ab response to protein antigens. There were no allo-hemagglutinins (group A). The patient had detectable, but very low titers of antibodies against polysaccharide antigens at the age of 11 yr, before vaccination. After vaccination with 23-valent nonconjugated vaccine at the age of 11 yr, titers rose and reached normal levels.

Mutations in the five known MSMD-causing autosomal genes (*IFNγR1, IFNγR2, STAT1, IL12B, IL12RB1*) were excluded in the three kindreds by means of genetic and immunological assays (unpublished data). Other patients with well-defined genetic defects were enrolled in our study, including patients with IL-12p40 deficiency, IL-12RB1 deficiency, IFN-γR1 deficiency, CD40 deficiency, and CD40L deficiency. Their genetic lesions are available upon request. Our study was conducted according to the principles expressed in the Helsinki Declaration and was approved by our Institutional Review Boards. An informed consent was provided by all patients studied, or by their parents, in the case of children.

Blood cell culture and stimulation. Whole blood samples were diluted 1:2 in RPMI 1640 (GIBCO BRL) and infected by incubation with live *M. bovis* BCG (Pasteur substrain), at a multiplicity of infection of 20:1, alone or with recombinant IFN-γ (5,000 IU/ml; Innokin, Boehringer Ingelheim) or recombinant IL-12p70 (20 ng/ml; R&D Systems). LPS (from *Salmonella minnesota*, 1 μg/ml; Sigma-Aldrich). TNF-α (20 ng/ml; R&D Systems), IL-1β (10 ng/ml; R&D Systems), phorbol 12-myristate 13-acetate (PMA) (10−7 M; Sigma-Aldrich) with ionomycin (10−7 M; Sigma-Aldrich) and supernatants were recovered after 14 and 48 h. Cytokine production was normalized according to the number of PBMCs in the individual tested. PBMCs were obtained from patients and healthy donors by centrifuging heparin-treated blood diluted 1:2 in RPMI 1640 on Ficoll-Paque Plus (GE Healthcare), followed by isolation in mononuclear cell medium. PBMCs were cultured in
RPMS 1640 medium (GIBCO BRL) supplemented with 10% heat-inactivated pooled FBS (GIBCO BRL), referred to as complete RPMI 1640. Cells (10^5 PBMCs/ml/well) were activated by incubation with phytohemagglutinin-P 1/700 (PHA, Bacto PHA-P; Becton Dickinson), alone or in combination with recombinant IFN-γ (5,000 IU/ml; Imukin, Boehringer Ingelheim) or recombinant IL-12p70 (20 ng/ml; R&D System) or IL-23 (20 ng/ml; R&D Systems), anti-CD3 antibody (10 ng/ml, OKT3, OrthoPharmaceutical) and LPS (from Salmonella minnesota, 1 μg/ml; Sigma-Aldrich) for 14 and 48 h.

Culture and stimulation of cell lines. Fibroblast cell lines were derived from dermal biopsy samples from patients and healthy donors and immortalized by transformation with a plasmid-containing SV40 large T antigen. We added 10^6 fibroblasts to DMEM (GIBCO BRL) supplemented with 10% (vol/vol) heat-inactivated FBS (GIBCO BRL) in each well of a 24-well plate. Fibroblasts were stimulated with TNF-α (20 ng/ml; R&D Systems), IL-1β (10 ng/ml; R&D Systems), or LPS (10 μg/ml; Sigma-Aldrich) plus ionomycin (1 μg/ml Sigma-Aldrich) and supernatants were recovered after 24 h of activation. Lymphocytes were immortalized with Epstein-Barr virus (EBV-transformed B cells) and cultured in complete RPMI 1640. EBV-transformed B cells were stimulated with mitomycin-C (10 μg/ml), an agent of TLR-7 and TLR-8 (provided by R. Müller, Pharmacia and 5M Innovative Properties Company, St. Paul, MN) for 24 h.

Genetic analysis. Genomic DNA was isolated by phenol/chloroform extraction from blood cells, which were washed in TE 10:1 buffer (10 mM Tris, 0.1 M EDTA, 0.5% SDS, and 10 mg/ml proteinase K) over-night at 37°C. DNA was extracted from EBV-transfected B cell lines or SV40-transformed fibroblasts with TRIzol (Invitrogen), according to the kit manufacturer’s instructions. We used 1–5 μg total RNA for direct reverse transcription with Oligo-dT (Invitrogen). PCR was performed using Taq polymerase (In Vitrogen), and the GeneAmp PCR System 9700 (Applied Biosystems). Primer sequences for the amplification of NEMO exons and cDNA are available upon request. PCR products were purified by centrifugation through Sephadex G-50 Superfine resin (GE Healthcare) and sequenced with the BigDye Terminator Cycle sequencing kit (Applied Biosystems).

Western blot and EMSA. For EMSAs, nuclear extracts were prepared by centrifugation through Sephadex G-50 Superfine resin (GE Healthcare) and sequences were analyzed on a 3100 ABI Prism Genetic Analizer by electrophoresis on a 3% stacking gel and 1.2% agarose gel, previously stained with ethidium bromide, and transferred to nitrocellulose membranes (Schleicher & Schuell). Membranes were blocked by incubation in 5% nonfat milk powder (Nestle USA) in 0.05% Tween-20 in PBS for 60 min at room temperature. NEMO was detected using rabbit polyclonal anti-NEMO antibodies (3328) and p18, gift from G. Courtois, INSERM, Saint Louis Hospital, Paris, France), rabbit antibodies against GAPDH (SC-25778, Santa Cruz Biotechnology, Inc.), rabbit anti-IκBα (C-21, sc-37, Santa Cruz Biotechnology, Inc.), and mouse antibodies against Stat-3 (Santa Cruz Biotechnology, Inc.). Membranes were washed several times in 0.05% Tween-20 in PBS, and antibody binding was detected by incubation with horseradish peroxidase–conjugated anti–rabbit secondary antibodies (GE Healthcare), using the ECL system (GE Healthcare).

Purification of peripheral T cells and monocytes. For T cell isolation, we depleted peripheral PBMCs of adherent cells and performed negative immunomagnetic depletion using a cocktail of antibodies against CD14, CD16, CD19, CD36, CD56, CD123, and glycophorin A (Macs; Miltenyi Biotech), according to the kit manufacturer’s instructions. The preparation was >95% pure, as assessed by flow cytometry with FITC-CD2 and PE-CD3 staining (BD Biosciences). Monocytes were isolated from peripheral PBMCs by negative immunomagnetic depletion, using a cocktail of antibodies against CD3, CD7, CD16, CD19, CD56, CD123, and glycophorin A (Macs; Miltenyi Biotech), according to the kit manufacturer’s instructions. Monocytes were >80% pure, as assessed by flow cytometry with PE-CD14 (BD Biosciences) staining.

Coculture of monocytes and T cells. We plated 10^5 monocytes per well in complete RPMI 1640 medium in 24-well plates (Nunc) and incubated them for 3 h at 37°C in an atmosphere containing 5% CO₂. The nonadherent cells were washed off and the adherent cells were incubated overnight with 400 μl of complete RPMI 1640. The following day, T cells were purified as described in a previous paragraph and 5 × 10^5 T cells were added to the monocytes. Selected wells were stimulated with a 1/700 dilution of PHA (Bacto PHA-P; Becton Dickinson), alone or in combination with recombinant IFN-γ (5,000 IU/ml; Imukin, Boehringer Ingelheim, France), recombinant IL-12p70 (20 ng/ml; R&D Systems), and recombinant IL-23 (20 ng/ml; R&D Systems). Supernatants were collected 14 and 48 h after activation.

Generation of dendritic cells from purified monocytes. MDDCs were obtained from purified monocytes as described in a previous paragraph. They were incubated for 2 h in complete RPMI 1640 medium at 37°C under an atmosphere containing 5% CO₂ to remove all nonadherent cells, thereby increasing purity. The adherent cells were cultured in complete RPMI 1640 in the presence of GM-CSF (50 ng/ml; R&D Systems) and IL-4 (10 ng/ml; R&D Systems). Optimal conditions were maintained by splitting these cultures every 2 d. Cells were collected on day 7, when the cultures contained >95% immature CD14+ (BD Biosciences) dendritic cells.

CD40 activation. Mouse fibroblastic L-cells transfected with the human ligand of CD40 (L-cell-hCD40L) were used to induce CD40 activation on MDDCs. Nontransfected L-cells were used for control cultures. L-cells were first treated with 10 μg/ml mitomycin (Sigma-Aldrich) in sterile 1 × PBS for 2 h at 37°C in 5% CO₂. L-cells were washed five to seven times in 1 × PBS to remove all trace of mitomycin. We then plated 10^6 L-cells per well in 24-well plates and incubated them for 2 d in complete RPMI 1640 at 37°C in an atmosphere containing 5% CO₂. We incubated 2 × 10⁵ MDDCs in complete RPMI 1640 with mitomycin-treated L-cells and recovered cell-free supernatants for cytokine analysis and MDDCs for cell surface phenotyping of CD40, CD80, and CD86 (BD Biosciences) by flow cytometry 24 h later. CD40 was activated on B cells with a mixture of soluble recombinant CD40L (500 ng/ml; Amgen) and IL-4 (100 IU/ml; R&D Systems). In brief, PBMCs were cultured for 5 d for proliferation (assayed by [3H]thymidine incorporation) assays and 12 d for the measurement of IgE production (assayed by ELISA) as previously described (45).

Flow cytometry. For the staining of NEMO in EBV-transformed B cells, fibroblastic cell lines, and highly purified monocytes and T cells isolated from PBMCs, we fixed 2 × 10⁵ cells with 4% paraformaldehyde (PFA, Fluka) in 1 × PBS and permeabilized the cells by incubation in 0.1% saponin (Sigma-Aldrich) in PBS. The cells were incubated with anti-NEMO (611306; BD Biosciences) antibody and the corresponding mouse IgG1 isotype (BD Biosciences), and primary antibody binding was detected by incubation with Alexa Fluor 488–labeled anti-mouse antibodies (Invitrogen). Cell surface phenotyping was performed by flow cytometry, in MDDCs incubated with L-cell or L-cell-hCD40L, using directly labeled antibodies. Dual-color fluorescence assays were performed with PE-CD11a.
(BD Biosciences) in combination with FITC-CD40, FITC-CD80, and FITC-CD86 (all from BD Biosciences). The respective isotype controls were performed with mouse PE-IgG2a (BD Biosciences) and mouse FITC-IgG1 (BD Biosciences). Fluorescence was analyzed with a FACSscan apparatus (Becton Dickinson), using CELLQuest software.

Cytokine analysis. Cytokine concentrations in supernatants were measured by two-sided sandwich ELISA, according to the kit manufacturer’s instructions: IL-12p70, IL-10, IL-6, IL-1β, IL-1β, IL-2, IL-4, IL-5, IL-6, IL-7, IL-8, IL-10, IL-12p70, IL-13, IL-17, TNF-α, IFN-γ, GM-CSF, G-CSF, MCP-1, and MIP-1β (Bio-Plex Suspension Array System).

Structural modeling of the NEMO oligomerization domain. The NEMO sequence encompassing the CC2-LZ region is not >30% of identity to any reference protein in the Protein Data Bank. We therefore used experimental data to search for comparable structure references. The NEMO oligomerization domain forms a globular trimer by equilibrium sedimentation with a hydrodynamic radius of 26.1 Å, as shown by gel filtration (30) and dynamic light scattering (unpublished data), and the CC2 and LZ coiled-coil subdomains interact by fluorescence polarization (30). Limited proteolysis in combination with mass spectrometry identified a tryptophan-sensitive site at residue Lys302, coinciding with the loop connecting the CC2 and LZ coiled-coils (unpublished data). Finally, the CD spectrum of the trimer showed it to contain 92% α-helix at 277 K (unpublished data). Based on these data, we used the HIV-1 gp41 ectodomain (PDB no. 1F23; reference 31) as a structure reference to provide atomic coordinates. The trimeric CC2-LZ domain of NEMO was modeled using the Insight II program (Accelrys, Inc.). The model was constructed by manually docking the Ca backbone of the CC2 coiled-coil (residues 260–292) with that of the LZ coiled-coil (residues 306–344) in an antiparallel manner. During docking, we looked for unfavorable and favorable contacts between the CC2 and LZ atoms, by calculating electrostatic and van der Waals interaction energies, using the Docking module of Insight II. The resulting energy-minimized model is shown in Fig. 2.

Immunofluorescence microscopy. MDDCs were allowed to differentiate and were CD40L-expressing fibroblasts, as previously described. MDDCs were immediately fixed upon activation, by incubation in 4% formaldehyde/PBS at 4°C for 20 min, followed by surface staining with mouse anti–human CD1a (BD Biosciences) at 4°C for 15 min. Cells were washed in PBS and incubated with goat Cy3-conjugated anti–mouse IgG antibody (Zymed Laboratories). For intracellular staining, MDDCs were permeabilized by incubation in 0.2% Triton X-100 (Sigma-Aldrich) in PBS for 15 min. Cells were washed twice in PBS, blocked by incubation with FcR-blocking reagent (Macs; Miltenyi Biotec) for 20 min, followed by surface staining with mouse PE-IgG2k (BD Biosciences) and mouse FITC-IgG1 (BD Biosciences), and were activated by CD40L-expressing fibroblasts, as previously described. The resulting energy-minimized model is shown in Fig. 2.

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We thank A. Munnich, A. Smahi, C. Hivroz, G. Courtois, R. Döffinger, and all members of the Laboratory of Human Genetics of Infectious Diseases for helpful discussions. We are particularly grateful to the families for agreeing to participate in this study. We thank T. Leclerc, M. Courat, and C. Bidal for excellent technical and secretarial assistance. A. Pienaar and L. Notarangelo thank the Centro per le Immunodifenzione primitive “Mario di Martino”.

O. Filipe-Santos was supported by Fundação para a Ciência e Tecnologia, Portugal; J. Bustamante was supported by the Fondation Schumlinger and INSERM. The Laboratory of Human Genetics of Infectious Diseases is supported in part by grants from the Schumlinger and BNP Paribas Foundations, the March of Dimes, and by EU grant QLK2-CT-2002-00846. S.M. Holland and M. Haverkamp acknowledge the support of the intramural program of the National Institute of Allergy and Infectious Diseases, National Institutes of Health. Studies by E. Vinolo, F. Agius, M. Wron, and A. Israel were supported by the Association pour la Recherche contre le Cancer and the Cancéropôle Ile de France. J-L. Casanova is an International Scholar of the Howard Hughes Medical Institute.

The authors have no conflicting interests.

Submitted: 9 January 2006
Accepted: 25 May 2006

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